SECOND EDITION

Handbook of Veterinary Nursing

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WILEY-BLACKWELL

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VETERINARY NURSING

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Preface

This second edition is intended to continue the aim that we set out to achieve with the first book: primarily to provide the veterinary nurse with a quick reference point for many of the nursing procedures he or she may come across in the clinical situation. We have updated chapters from the first edition and, most notably, added an entirely new chapter in Section 1 in response to the changes to the practice of veterinary nursing over the past decade.

In the 9 years since writing the first edition, a lot has happened not only in our personal and professional lives, but more specifically to veterinary nursing which has made writing this edition more of a challenge. The 'professionalisation' of our vocation has driven adjustments in legislation, introduced a register of veterinary nurses and been responsible for the way veterinary nurses now learn their trade. Changes are happening so rapidly at the moment, that by the time of publication of this book, it is likely that significant developments will have taken place that will have an impact on how veterinary nurses practise. A prime example of this is the proposed disciplinary scheme for veterinary nurses which may well already be implemented.

In writing this edition, we have tried to address these issues and provide readers with a practical and straightforward guide which they can use to continue to develop their clinical practice.

Now more than ever, veterinary nurses are having to respond to the changing culture of veterinary nursing. Their day-to-day practice is undoubtedly changing; the general public's demand for transparent and high-level veterinary care is leading to increased responsibilities and scope for veterinary nurses and thus the need for veterinary nurses to clarify their role and rationalise the care they give their patients.

As with the first edition, it is not intended that this handbook is taken as a definitive text, but rather to be used as a quick reference source for all nurses at all stages of their career. We hope that this format allows easy access to clinical procedures and helps veterinary nurses when they are faced with difficult professional decisions or dilemmas.

This book is intended to be a guide to quality care and skills. However, none of the procedures should be carried out without appropriate instruction first and then only under the direction of a veterinary surgeon. Veterinary nurses must be familiar with and work within the RCVS Guide to Professional Conduct and keep up to date with the ongoing amendments and reviews to the Veterinary Surgeons Act 1966, Schedule 3 which allows non veterinary surgeons to undertake certain acts of veterinary surgery.

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SECTION 1

CONTEMPORARY VETERINARY NURSING

INTRODUCTION

This section looks into the essence of modern day veterinary nursing by examining a brief history of veterinary nursing and considering how this heritage has had an influence on the way veterinary nurses practise in the 21st century. The Registered Veterinary Nurse's (RVN's) responsibilities as a professional practitioner are identified in Chapter 1 and guidance given to help RVNs work their way through some of the issues that they will face in their day-to-day work. The concept of the nursing process and how this may be applied to the veterinary patient is covered in Chapter 2. Using this structure, the importance of a thorough assessment is highlighted through the remaining chapters in order to provide a more holistic approach to patient care. A framework of assessment is employed in the form of a model of care (the ability model) which has been developed specifically for veterinary nursing.

Chapter 1

Present-day Veterinary Nursing in the UK

In the relatively short time that veterinary nursing has been a recognised vocation, there have been developments in society which have had an impact on the way veterinary nurses practise in the 21st century. A brief review of the history of veterinary nursing since its formal inauguration 50 years ago can help us begin to appreciate how our heritage and, to some extent, external influences have contributed to the way we currently practise. Reflecting on the past and present helps us to consider the future of veterinary nursing practice and should enable us to develop our profession to benefit patients, veterinary nurses and employers. The overarching theme behind all of the developments during this time has been to improve the relatively lowly status of veterinary nurses and ultimately achieve professional status and, with this, recognition.

A BRIEF HISTORY

Before 1961 veterinary nursing did not exist as a formally recognised career. There are accounts of people working as lay assistants and 'canine nurses' before this time, but the efforts of veterinary members of the British Small Animal Veterinary Association (BSAVA) were needed to convince the Royal College of Veterinary Surgeons (RCVS) that, if they were trained formally, these people could be of huge benefit to the veterinary profession. The RCVS introduced a training and qualification scheme which allowed the first Registered Animal Nursing Auxiliaries (RANAs) to qualify in 1963 (Pullen & Gray, 2006). This saw the start of a movement to create a support system and veterinary nursing community network for members of the vocation. A group of forward-thinking veterinary surgeons were instrumental in the setting up of the British Veterinary Nursing Association (BVNA) in 1965, which acted as a representative body for veterinary nurses.

Up until the late 1970s in the UK, the use of the title 'nurse' was protected by law and could only be used by nurses (midwives and health visitors) registered with the General Medical Council. Following a change in legislation to release the title from its protected status, the title nurse could be used by other vocations such as veterinary, dentistry and nursery workers (RCN, 2003) and the RANA title was changed to veterinary nurse (VN) in 1984.

A hugely significant development to the role and job prospects for veterinary nurses came in 1991 with the Amendment to Schedule 3 of the Veterinary Surgeons Act (1966). Prior to 1991, veterinary nurses were not recognised

legally as being any different from other members of the public. It meant that only a veterinary surgeon was authorised to carry out medical treatment or surgery to animals (although animals' owners were allowed to give some minor treatment). Following the amendment, RCVS-qualified, listed veterinary nurses were formally recognised and permitted to undertake certain procedures which were not permitted by lay staff or members of the public.

The way veterinary nurses were trained and educated underwent significant modifications and developments in the mid 1990s with the conversion of the veterinary nursing certificate qualification to a National Vocational Qualification and the launch of other accredited nursing qualifications such as the Diploma in Advanced Veterinary Nursing and the introduction of full-time university degree programmes in veterinary nursing. With these educational changes came a body of veterinary nurses with specialist and higher-level qualifications and the trend developed towards more widespread teaching of concepts and theories used in human nursing.

As part of the continued drive for veterinary nursing to achieve a higher status and become a profession in its own right, the first RCVS VN council was formed in 2002, comprised of elected veterinary nursing members. In 2007 the non-statutory RCVS Register of Veterinary Nurses was introduced which meant that all VNs qualifying with the RCVS after 2003 achieved automatic registration status. In a further bid to achieve professional status, the case for a system of regulating veterinary nurses has been proposed to help ensure the protection of the public through the mechanisms of regulation. At the time of writing, the RCVS disciplinary system for veterinary nurses is due for implementation by the end of 2010.

Increased recognition and status

Popular television series such as the BBC's fly-on-the-wall documentaries Animal Hospital (1994–2004) and Super Vets (2006–2007) featured the veterinary profession at work and have undoubtedly raised the profile of veterinary nursing as a career in its own right and helped to increase the general public's awareness of the role of the veterinary nurse.

The combination of events of the past 47 years, and the vocation's drive towards professionalisation in the past 20 years, means that veterinary nurses can now enjoy improved career opportunities, increased recognition and improved employment status. Veterinary nursing practice is undoubtedly changing and so too is the VNs' relationship with 'professional' issues such as the law, the RCVS and their ethical duties to patients and their owners. To achieve and maintain professional status RVNs must continue to respond to the changing environment and modify their working practices accordingly. They must also continue to be proactive in developing the profession.

THE REGISTERED VETERINARY NURSE AS A PROFESSIONAL PRACTITIONER

With greater responsibility comes accountability. The professional and legal responsibilities of RVNs extend not only to their animal patients, but to the

animals' owners, their professional colleagues and society at large. Professional responsibility, however, is a relatively new concept for veterinary nurses and so it is essential that RVNs ensure that they are aware of the legal, professional and ethical ramifications of any decisions they make or actions they take.

A professional person is legally *accountable* for his or her actions in four main areas (Dimond, 1995):

- to society through the criminal courts;
- to the client through the civil courts;
- to the profession through the Professional Investigation Committee (Royal College of Veterinary Surgeons);
- to his or her employer through contracts of employment and disciplinary procedures.

Accountability

Accountability forms an integral part of being a professional and relates to the legal, professional and ethical duties that we have towards other people. There may be times when the RVN is faced with a dilemma about how he or she should act in a particular situation. Responsibilities to patients, patients' owners, the directing veterinary surgeon, employer and professional body can lead to situations where conflicts have to be balanced and it can be difficult for the RVN to know how to act. The RVN must be able to make and justify the correct decision or action.

LEGAL RESPONSIBILITIES

The law deals with maintaining and defining sets of rules and the systems for their enforcement. It is easy to think of the law as being concerned solely with crime and punishment, but in reality a great deal of law is concerned with the rights and responsibilities of individuals and companies towards each other.

Responsibility of the veterinary nurse to society through the criminal courts

Criminal law is concerned with acts which are forbidden in order to protect society and individuals from harm. Someone who commits a crime commits an offence against the state and the state may prosecute that person. A conviction can result in the accused serving a prison sentence or in some cases paying a fine and/or performing community service – the level of punishment depends on the particular crime and any mitigating factors. Criminal law is not just about things like murder, assault, theft and traffic offences. There are a number of laws which carry a criminal sanction which directly affect the work veterinary nurses do; these include:

- Veterinary Surgeons Act 1966;
- Health and Safety at Work etc Act 1974;

- Medicines Act 1971;
- Misuse of Drugs Act 1971;
- Dangerous Dogs Act 1991;
- Data Protection Acts 1984 and 1999;
- Animal Welfare Act 2006;
- Protection of Animals Acts 1954 and 1964;
- Animals (Scientific Procedures) Act 1986;
- Public Interest Disclosure Act 1998.

Veterinary Surgeons Act 1966

Section 27 of the Veterinary Surgeons Act 1966 (VSA) defines the practice of veterinary surgery as

> *… the art and science of veterinary surgery and medicine and, without prejudice to the generality of the foregoing, shall be taken to include –*
>
> **(a)** *the diagnosis of disease in, and injuries to, animals including tests performed on animals for diagnostic purposes;*
> **(b)** *the giving of advice based upon such diagnosis;*
> **(c)** *the medical or surgical treatment of animals;*
> *and*
> **(d)** *the performance of surgical operations on animals.*

Section 19 of the Act restricts the practice of veterinary surgery to individuals '*… registered in the register of veterinary surgeons or the supplementary veterinary register …*' and certain exempted categories of people and procedures set out in Schedule 3 to the Act.

Schedule 3 can be and has been changed from time to time by regulations. At the time of writing the current version of Schedule 3 of the VSA is contained in a Statutory Instrument (SI 2002/1479) which sets out the treatment and operations which may be carried out by '*unqualified persons*'. These include:

- minor medical treatment given to an animal by its owner, or a member of the owner's family or the owner's employee;
- certain specified procedures on animals used for agriculture by their owner or a person employed in looking after them;
- the rendering in an emergency of first aid for the purpose of saving life or relieving pain and suffering;
- certain specified procedures which may be done by anyone over the age of 18 years;
- certain procedures which can be carried out in specified circumstances by persons of 17 years if they are undergoing instruction in animal husbandry;
- provisions relating to veterinary nurses and student veterinary nurses which will be considered in more detail.

Paragraphs 6 and 7 of Schedule 3

Paragraph 6 of Schedule 3 permits:

> *Any medical treatment or any minor surgery (not involving entry into a body cavity) to any animal by a veterinary nurse if the following conditions are complied with, that is to say –*
>
> **(a)** *the animal is, for the time being, under the care of a registered veterinary surgeon or veterinary practitioner and the medical treatment or minor surgery is carried out by the veterinary nurse at his direction;*
> **(b)** *the registered veterinary surgeon or veterinary practitioner is the employer or is acting on behalf of the employer of the veterinary nurse; and*
> **(c)** *the registered veterinary surgeon or veterinary practitioner directing the medical treatment or minor surgery is satisfied that the veterinary nurse is qualified to carry out the treatment or surgery.*

'*Veterinary nurse*' is defined as meaning a nurse whose name is entered in the list of veterinary nurses maintained by the RCVS.

VNs must be aware that failure to satisfy all the various conditions constitutes a criminal offence. While they may carry out any *medical* treatment, they may only carry out minor surgery which does not involve entry into a body cavity, but in either case, they may only do so if all three conditions in (a), (b) and (c) are met.

Paragraph 7 sets out provision for student veterinary nurses, defined as '*a person enrolled under bye-laws made by the {RCVS} for the purpose of undergoing training as a veterinary nurse at an approved training and assessment centre or a veterinary practice approved by such a centre.*' It contains similar wording to Paragraph 6 permitting:

> *Any medical treatment or any minor surgery (not involving entry into a body cavity) to any animal by a student veterinary nurse if the following conditions are complied with, that is to say –*
>
> **(a)** *the animal is, for the time being, under the care of a registered veterinary surgeon or veterinary practitioner and the medical treatment or minor surgery is carried out by the student veterinary nurse at his direction and in the course of the student veterinary nurse's training;*
> **(b)** *the treatment or surgery is supervised by a registered veterinary surgeon, veterinary practitioner or veterinary nurse and, in the case of surgery, the supervision is direct, continuous and personal; and*
> **(c)** *the registered veterinary surgeon or veterinary practitioner is the employer or is acting on behalf of the employer of the student veterinary nurse.*

So the essential difference between what VNs and student VNs are permitted to do is that

- the treatment or surgery must be carried out in the course of the student's training;

- the student must be supervised by one of the specified categories of people, and, in the case of surgery, the supervision must be direct, continuous and personal.

Failure to comply with these conditions is a criminal offence. It is essential that VNs and student VNs keep the limitations in mind and be prepared to insist on their being complied with. If a student VN were, for example, to carry out surgery without '*direct, continuous and personal*'supervision from an appropriate person, it is the student who would be breaking the law, not the person who has failed to supervise adequately.

All VNs must be sufficiently familiar with and comply with relevant legislation and understand what it means in practice. The Government website www.opsi.gov.uk contains copies of UK acts and delegated legislation from 1988 onwards.

Responsibility of the veterinary nurse to the client through the civil courts

Civil law is concerned with relations between individuals and/or businesses. It differs from criminal law in that the disputes between individuals are not considered matters of public concern. In civil cases the claimant is often looking for financial compensation (or 'damages') for a loss suffered.

In the event of a veterinary nurse causing harm to a client's animal, the client might decide to sue for compensation for the damage or loss they have suffered. Usually the claim would be made against the owners of the practice rather than against a nurse who was just an employee. If a nurse employee was sued directly, then the practice should be *vicariously liable* for his or her acts provided he or she acted in the course of his or her employment. Veterinary practices are required to carry third-party insurance for the protection of the public.

The two areas of civil law that are most relevant to veterinary nurses are claims for breach of contract and the tort of negligence.

Contract

A contract is an exchange of promises in which one party agrees to do something in exchange for something else. In the veterinary context, owners enter into a contract with the practice when they bring their animal in: the practice agrees to examine the animal and the owner agrees to pay. The terms of the contract may then be extended to cover treatment.

The starting point is the identity and capacity of the person bringing the animal in. Are they its owner? Can they consent to treatment? Can they agree to pay? If a child brings an animal in, it would be prudent to make sure that a parent or guardian agrees to pay and consents to any treatment. If an adult client is the owner's friend, spouse/partner or relative, one should be sure not only that they or the owner agree to pay, but also that the owner consents to the treatment or has authorised the other person to do so on their behalf. Carrying out treatment which the owner did not agree to could lead to a claim against the practice.

Some practices have standard written terms of business on which they agree to work; others may simply do so on an oral instruction. The advantages to a practice of putting its terms of business in writing come down to trying to ensure that it gets paid and does not get sued unnecessarily. Putting things in written form and getting the client to sign reduces the scope for argument over what the arrangement is between the practice and the owner; for example, an owner might say that they thought that an initial consultation was free, or that they did not expect to be charged for something, or that they did not expect to be charged so much.

If there is no written agreement, it is much harder to prove what was agreed, as it is one person's word against another. In such a case, good contemporaneous notes recording what was agreed can be invaluable.

Whether an agreement is written or oral, the law implies terms into the contract. As a business arrangement, the owner can expect that the work will be carried out with reasonable skill and care.

Consent to treatment should ideally be in writing, signed by the owner. In the human context, the patient (or someone on his or her behalf) gives consent to what would otherwise legally be an assault. It is different with animals. They cannot give consent themselves and the law considers them as someone's 'chattels', or personal property. The purpose of consent in the veterinary context therefore supplements the business agreement between the practice and the owner by recording what the practice has agreed to do and what the owner has agreed to pay for.

Problems can arise when owners did not consent (or say that they did not consent) to particular services being performed, or when the results of the service they receive are not what they thought they would be. To a great extent, this is a matter of managing expectations. As the outcome of a patient's treatment is often uncertain, it is important that the client knows what treatment their animal will receive and the risks involved, and that they agree to this. Owners should not be able to claim that had they known a particular fact, they would not have agreed to something. For example, if the client appeared to think that a procedure would be performed by a veterinary surgeon rather than a veterinary nurse, it might be prudent to ensure that they were made aware of the fact and consented to it.

Consent can be given verbally: there will inevitably be occasions when an urgent telephone call is made to a client for instructions. But as with all verbal agreements, there is no proof that consent was given. Where possible, consent should always be in writing and signed by the owner. If this is not possible, the person speaking to the client should make a good contemporaneous note of the conversation and the instructions, and make sure that it is kept on file. There may be circumstances where a practice might wish to think very carefully before going ahead even then.

English law does not recognise that loss of, or damage to, chattels causes distress to their owners for which compensation is payable. An owner must show not that he or she is distressed, but that he or she has suffered a financial loss arising from a breach of the contract with the practice. This is clearly easier for someone to show in the case of a valuable pedigree animal with breeding potential.

Negligence

Negligence is a breach of the duty to take care. The tort of negligence can exist independently to a contractual relationship. For a claim of negligence to hold, three fundamental criteria must be established:

- a duty of care exists between the parties;
- there has been a breach of the duty of care;
- the breach has caused reasonably foreseeable harm.

In the context of a veterinary practice, it can be assumed that it owes a duty of care towards the owners of its patients with whom it has contracted.

To determine if a breach in the duty of care has occurred, it is necessary to clarify the standard of care expected. The classic test in the case of skilled professionals is the so-called Bolam test. This is that the person owing the duty of care must have acted in accordance with a practice and level of skill accepted as proper by a responsible body of his or her fellow professionals. In the context of a typical veterinary practice, a claim for negligence would probably be made against the practice rather than an individual because it was the practice to whom the owner entrusted his or her animal, and the standard of care they would expect would be that they should receive from a reasonably competent veterinary surgeon.

The final criterion for a successful claim of negligence is to determine if the breach in the duty of care has caused harm which was reasonably foreseeable. In other words, would the damage have occurred 'but for' the negligence? This can be arguable, particularly in medical (and veterinary) situations, where treatment by even the most experienced practitioner does not guarantee a 100% successful outcome.

Responsibility of the veterinary nurse to the profession

The RCVS is the governing body of the veterinary profession and one of its primary functions is to protect the public and their animals by setting out the professional standards required of its members. Professional standards are set out in the Guide to Professional Conduct and the RCVS takes responsibility for the professional regulation of its members and it manages this through its disciplinary procedures. A non-statutory register of veterinary nurses came into existence in 2007 with mandatory registration of all VNs qualifying after 2003 and voluntary registration for those who were qualified and listed prior to 2003. (Prior to this, the RCVS held a 'list' of qualified veterinary nurses.) As yet, there is no statutory registration for veterinary nurses, so Schedule 3 of the 1991 Amendment Order of the VSA, allows RCVS listed as well as registered VNs to undertake procedures covered by the Act. At the time of writing, the RCVS has no power to remove incompetent or criminal VNs from this list or from the register, however, disciplinary mechanisms for registered veterinary nurses are due to come into force in 2010.

Once this system is in place, any complaint about a registered veterinary nurse will be considered by the RCVS to determine if it constitutes serious professional misconduct. In the event of serious professional misconduct, the

VN disciplinary committee will decide on the action to be taken. Action could include the veterinary nurse being suspended for a fixed period or removed from the Register.

Continuing professional development

One of the requirements of being a professional practitioner is to keep up to date with developments in the profession and foster a reflective and lifelong approach to learning. The RCVS stipulates that RVNs are required to maintain their skills and standards of professionalism by means of continuing professional development (CPD). Currently, RVNs are required to undertake and record a minimum of 45 hours of CPD over a 3-year period. This may take the form of formal lectures and seminars but can also include reading articles, textbooks and carrying out one's own research into areas of interest. A record should be kept of any CPD activity along with some evidence of reflection and a statement on what learning or improvement in practice has taken place.

Responsibility of the veterinary nurse to the employer through contracts of employment

Employees have certain responsibilities to their employers – and vice versa – and these should be detailed in contracts of employment and disciplinary procedures. The disciplinary procedure is one way that an employer can raise concerns that something that an employee is (or is not) doing is unacceptable. An employer may initiate disciplinary procedures if he or she feels that an employee has breached some aspect of the contract or behaved in an unacceptable way. Disciplinary procedures may include formal verbal warnings, formal written warnings and termination of employment. Examples of activities which might be in the contract of employment, include the employer's policy on the following.

- Timekeeping.
- Absence.
- Health and safety.
- Personal appearance.
- Actions that might be seen to be of 'gross misconduct'.

These are usually serious enough to be a breach of contract between the employer and employee and which could result in instant dismissal. Examples of acts of gross misconduct could include:

- theft;
- serious misuse of an employer's name;
- bringing the employer into serious disrepute;
- incapability to work due to alcohol or illegal drugs;
- causing damage or injury through negligence;
- a serious breach of health and safety rules;
- a serious breach of confidence;
- bullying or physical violence.

Fig. 1.1 Gibbs' reflective cycle.

THE REFLECTIVE PRACTITIONER

Within veterinary nursing integration of theory and practice is essential. This can be achieved by reflecting on everyday experiences. Experiential learning is an essential in a job which varies on a daily basis. Reflection occurs on a daily basis but not always using a framework. A student nurse quickly learns that removing an aggressive cat from a kennel requires more than just a thin towel. By taking time to analyse a situation that has occurred, the individual calls on prior knowledge (or experience) in order to make sense of the situation. This can then lead to an improvement in practice where appropriate. There are several frameworks that can assist in the process of reflection. Gibbs' (1988) reflective cycle consists of a series of questions to guide the reflective practitioner (Fig. 1.1).

- **Stage 1: description – what happened?** It is important to be able to describe the event thoroughly. What was the situation, who was involved, what was your role and what was the result? Often writing this down helps to start to make sense of what might initially seem a confusing situation.
- **Stage 2: what were you thinking or feeling?** Self-awareness is an important factor in being a reflective practitioner. How did the situation affect you, how did it make you feel? How do you think the other people involved were feeling? How do you feel about the outcome? What are your thoughts now about the situation?
- **Stage 3: evaluation and analysis.** What was good or bad about the situation? What did not go as well as expected? How did you make sense of the situation? Breaking the situation or incident into components may help with trying to analyse the situation. Refer back to prior knowledge – what happened in a similar situation?

- **Stage 4: conclusion.** What alternatives did you have? Having explored the situation you should now have information on which to base your judgement of the situation.
- **Stage 5: action plan.** What would you do differently next time? Now you have analysed the situation, what have you learnt and would you change what you would do or how you would act next time?

A simpler but just as effective model of reflection is Rolfe's (Rolfe *et al.*, 2001) framework for reflective practice. It consists of three questions (Table 1.1).

Table 1.1 Rolfe's framework for reflective practice.

What?	So what?	Now what?
What was the situation? Describe what happened, what was good or bad	So what was important about what happened? How did I feel and what sense did I make of the situation?	Now what could I do? What will I do differently next time?

Reflecting in this way helps the individual to learn constructively from both everyday experiences and more critical incidences that may occur in the practice.

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Chapter 2

The Nursing Process

THE ROLE OF THE VETERINARY NURSE

Most veterinary nurses understand that delivering high-quality and appropriate care to their patients is central to their role. With recent changes regarding the regulation of the veterinary nurse, it is important that nurses recognise their responsibility, not only in providing the care for their patients, but also to the owners of patients. Along with the increased responsibility that regulation brings, nurses must expect to be held accountable for their actions and the consequences that result when failing to recognise this.

The changing focus of veterinary nursing care

Veterinary nursing has traditionally followed a biomedical model approach to the teaching of veterinary nursing and a practical approach to patient care. The biomedical model is the basis on which many modern western medical professions teach and practise medicine. It is based on the premise that illness is due solely to malfunctioning physiology and/or anatomy and consequently patients are defined by their illness or medical condition. Treatment and care are based around the patient's physical disorder(s) and pathophysiology. Whilst the biomedical model follows a systematic approach to the anatomical, physiological and biochemical disorders associated with illness, its suitability for diagnosis and treatment of people has been rejected for a number of years by various medical professionals because it focuses on the disease as a problem to be fixed and takes no account of many of the other factors that might contribute to a person's physical and mental health status.

As a result, various paramedical professions have developed and adopted other models of care through which to focus their approach to patient care. Most notably, the nursing profession deduced that the biomedical model failed to foster a holistic approach to nursing of patients and that it was not the most appropriate way to structure nursing care. Since the 1980s the nursing profession began a widespread move away from the biomedical model, developing alternatives which took into account other factors, such as the patient's individuality, social factors and the interaction that occurs between the patient and the healthcare professional.

With the introduction in 2006 of nursing models of care and care plans into the RCVS veterinary nursing syllabus, the focus of veterinary nursing care

began the shift towards a more holistic approach to care delivery for veterinary patients by veterinary nurses.

INTRODUCTION TO NURSING THEORY

The terms *nursing process*, *nursing diagnosis*, *models of care* and *care plans* refer to separate concepts and frameworks used in nursing, but which are inextricably linked. To illustrate, a *care plan* is a written outline of the nursing actions systematically set out for a particular patient. To be able to devise a care plan, the nurse needs to have made a *nursing diagnosis*. To be able to reach a nursing diagnosis the nurse needs to have assessed the patient. A *model of care* helps the nurse assess the patient systematically and thoroughly, and subsequently can be used to identify the nursing actions required. All of this, when put together successfully, becomes the *nursing process*.

The nursing process is a problem-solving approach to caring for an individual patient. It involves assessing the patient's needs, identifying the problems which require nursing intervention and then planning and implementing the appropriate care required. The next stage in the process is to evaluate the effect of the nursing care provided. It is a cyclic process that may be repeated many times throughout the patient's period of hospitalisation. The application of the nursing process in veterinary nursing will be explored later in this chapter.

History

Florence Nightingale, often quoted as the founder of modern-day nursing, developed her own form of the nursing process. This was based on linking the environment to the health of the patient. Environmental issues such as physical factors (heating, light, cleanliness etc.), as well as the psychological and sociological environment, were seen by Nightingale to have a great impact on the health of the patient (Sibson cited in Peate, 2005).

The biomedical model was adopted by the veterinary profession, but the nursing profession deduced that the biomedical model failed to foster a holistic approach to nursing of patients and that it was not the most appropriate way to structure nursing care.

The nursing process idea was introduced into the syllabus for general (human) nursing in 1977. Up until then, nursing focused on patient care being broken down into a set of tasks which led to the nursing care being fragmented and nurses only being able to initiate very superficial nurse – patient relationships. The introduction of the nursing process led to the patients being treated more as a whole person rather than a collection of unit tasks.

> *Care of patients was always planned long before the phrase 'nursing process' was coined. The fundamental difference, however, is that the focus of care has changed from tasks to patients.*
>
> Mike Walsh (1997)

The general concept is that the *nursing process* is the '*way one thinks like a nurse*' (Murray and Atkinson, 1994, p1). Nursing theorists propose that experienced, expert nurses think automatically and act in a structured way, albeit without conscious thought, but if we were to unpick the phases of their thinking and action, it would amount to a clearly identifiable set of stages. The general consensus is that by identifying and teaching these stages, nurses are in a better position to deliver a consistently high standard of care. The nursing process does not necessarily detail *what* is done in each stage or *how* (this is done with various frameworks such as models of care and care plans) it merely outlines the stages involved. It moves away from the biomedical model approach to nursing towards a more holistic, patient-focused approach to patient care and rehabilitation.

The nursing process

The concept of the nursing process has been central to professional nursing practice in order to provide organised and holistic care to patients. By delivering nursing care in such a structured and systematic approach, it is hoped that the nursing care given is thorough, incorporating the individual patient requirements, rather than focusing predominantly on the patient's physical needs.

The nursing process provides a structure to the procedure of planning the care for the patient. This ensures the patient is assessed in such a way as to highlight issues which may affect the way care is given. Once the patient has been assessed, the nurse plans how the care might be carried out. Following the planning stage, the care plan that has been have drawn up is then implemented. Evaluation is then important in order to assess the effectiveness of the care that has been given.

The nursing process is traditionally divided into four stages (Fig. 2.1) but should be a cyclic process with continuous assessment and evaluation during the patient's stay in hospital, and often continuing when they go home if aftercare is required. A fifth stage, nursing diagnosis, was late introduced after assessment. The nursing diagnosis differs from medical diagnosis in that it emphasises the individual who is interacting with the environment and whose health status requires nursing intervention.

THE NURSING PROCESS IN VETERINARY CARE

Knowledge of caring for veterinary patients is usually gained from working with and observing the more experienced nurses and veterinary surgeons within the practice. Training courses may teach in bullet point form what to do with, for example, a diabetic dog. However this is very much a disease-orientated approach as seen with the medical model of care, meaning that care is centred around the animals' disease or malfunction of a body system rather than the whole animal. Everyone has, at some time, referred to their patients by what is wrong with them, '*the fracture in kennel 5*' and '*the bladder*

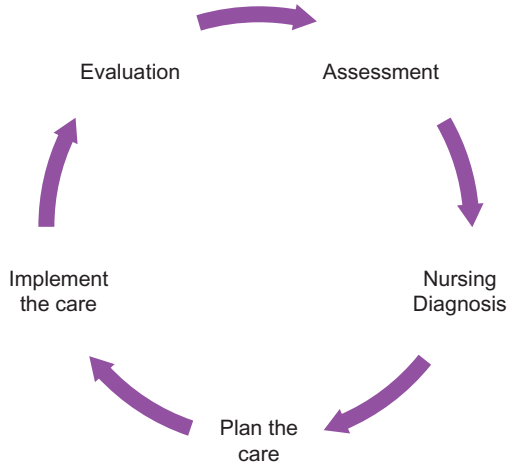

Fig 2.1 The nursing process.

cat in kennel 3', rather than considering the whole patient and the patient's needs. With the medical model, it is the physician who dictates the type of care the patient receives, with the nurse carrying out the orders as a series of tasks to deliver the care. In the traditional role of the nurse being the doctor's 'handmaiden', this type of care would not be questioned.

The following case is typical of the type of patient veterinary nurses care for on a day-to-day basis.

Dexter Smith is a 4-month-old entire male domestic longhair cat. He has been admitted to the hospital for investigation of intermittent vomiting, diarrhoea, inappetance and lethargy over several days. The veterinary surgeon has requested that he is kennelled for observation and preliminary blood tests before any further, more invasive investigation of the problem takes place.

How is the care planned for this patient?

When caring for a patient the VN will carry out the veterinary surgeon's requests but also contribute to the general well-being of the patient, e.g. grooming or encouraging the patient to eat, spending extra time with a patient that is uncomfortable or afraid. These are aspects of nursing that are not normally noted in the care requirements. When nursing a patient the emphasis maybe on treating the illness and administering the medication but the extra time spent in ensuring the patient is comfortable or able to move around is just as important. The term holistic care encompasses this way of nursing the whole animal rather than just its disease. So in fact, veterinary nurses have been carrying out the nursing process to a certain extent already – just not in a particularly structured or formal way. The lack of structure to the process may mean some aspects of caring for a patient are missed.

When Dexter is admitted to the ward, the VN may ask the veterinary surgeon some further questions about the patient. Calling the owner may also be an option to ask them some questions about Dexter. Carrying out a physi-

cal examination and observing Dexter in the kennel may also be performed. By completing any of these procedures the VN is carrying out an **assessment** to a greater or lesser extent.

From the brief assessment carried out and the results of the blood test and physical examination, the VN may conclude that the cat is showing early signs of dehydration and may be at risk of hypovolaemia if he is left untreated. He is not washing himself and the fur around his rear end is getting matted and covered in diarrhoea. He is not responding to the stroking or the nurse talking to him, and his demeanour is depressed. The continued lack of food will result in weight loss and his recovery from whatever physiological disorder is going on will take longer. The VN is making decisions on nursing care and is making a **nursing diagnosis**.

The VN needs to get him eating and drinking and feeling better about life. Grooming and tidying up his matted, faeces-covered rear end will also make him feel better and is essential for preventing further infection. Further nursing interventions will include trying to get Dexter to eat something by encouraging him, and perhaps aiming to monitor his urine output. The VN may decide to obtain his vital sign parameters and weight every day and speak to the veterinary surgeon about repeating a packed cell volume/total protein (PCV/TP) blood test the following day. This is **planning** Dexter's care.

At this stage the VN will be carrying out the planned nursing interventions. This will include tempting him with small amounts of warmed, aromatic food, stroking him and talking to him to try and make him feel better and encourage him to eat. The temperature, pulse and respiratory rate are recorded and he is weighed each day. This is **implementing** the planned nursing care.

Later on in the day, Dexter has vomited a small amount of yellow, frothy fluid. He has yet to urinate and despite all efforts, he is still not eating. He seems to be looking more depressed and is starting to show some clinical signs of dehydration. Dexter is not showing any signs of improvement and his condition appears to be getting worse. Further discussion with the veterinary surgeon takes place to decide what the next step might be. This is **evaluating** the nursing care.

The above scenario helps to explain that experienced nurses are already applying the nursing process. By combining nursing knowledge, experience, skill and judgment, Dexter receives a good level of nursing care. Student nurses are still building on their experience and knowledge and do not naturally nurse by intuition. This is where using a structured model of care ensures nursing care can be given equally by students and qualified nurses. A model of care will also guide the nurse's actions, ensuring a more holistic approach is carried out.

The assessment phase

The assessment phase is the systematic collection, organisation and analysis of information about an individual patient. Assessment is always the first stage, but is revisited frequently throughout the interaction with the patient to ensure the most appropriate care is being provided at any one time. Each

stage of the nursing process is of equal importance to its overall effectiveness of improving patient care. If one stage is skimped upon, this will be reflected in the quality of the next. This is particularly important for the assessment stage as it forms the basis for the remainder of the nursing process. The assessment phase must clearly establish the individuality of the patient, only then can effective care be delivered. It is important to collect appropriate information on the patient; this may be done by the VN's own observations, plus information from the client and other members of the team.

During this stage it is important to identify the patients' *actual* problems at the time of admission but also to note any *potential* nursing problems that may develop as a result of the existing problems. Some of these problems may be linked to specific medical problems whilst others may be more specific to that individual, involving psychological or social status. It is important to collect the information systematically and write it down.

Gathering the right type of information about a patient is very important. It sets the scene for any action to be taken. If the wrong information is collected at assessment it follows that the wrong action may then be taken. If insufficient information is collected, inadequate action may be taken.

Carrying out the assessment

There are several methods documented that may be used to carry out the assessment. The nursing process, after all, says VNs must assess but not what VNs should assess. In order to deliver the nursing process, a framework is required and this comes in the form of a model of care.

The SOAP model of assessment is frequently used in veterinary medicine, and focuses around the body systems (Table 2.1). Although this method of assessment was originally documented by Hildegard Peplau in her developmental model of care for psychiatric patients, it tends to be more generalised and not cover specific nursing interventions.

Nancy Roper, Winifred Logan and Alison Tierney developed a model of nursing in which the assessment process focused on the basic activities of

Table 2.1 The SOAP model for assessment.

S – Subjective What is wrong with the animal/patient. For example this might be right fore lameness, acute abdominal pain, chronic vomiting etc. It usually includes relevant history obtained from the owner/patient. May also state goals to be achieved (reduction of pain, reduction in lameness etc.).
O – Objective Results from a thorough clinical examination and diagnostic tests.
A – Assessment Includes differential diagnosis based on the subjective assessment and objective data.
P – Plan Plan of treatment/action required in order to progress and hopefully achieve the goals stated in the subjective part.
This format may also be extended to include carrying out the plan and evaluating the plan **SOAPIER:** **Implement** the plan **Evaluate** the effectiveness of the plan **Reassessment** or review of plan

living. It sought to identify which activities the patient was able to do and those for which they required assistance (from nursing personnel). Dorothea Orem developed a self-care model of nursing where the assessment focuses on what the patients can do for themselves. A model of care for veterinary nursing is described later in this chapter. It is currently under development but it is hoped that it will be adopted by veterinary practices.

Collecting information

Using a number of different methods, the nurse gains information (assesses) the patient. The main sources include:

- **the client** (the person who knows the most about the animal);
- **the patient** (observation is essential in nursing the veterinary patient who has other ways of communicating other than language);
- **significant others** (the admitting vet or nurse, nursing colleagues, animal care assistants, students).

The information is obtained using various methods:

- interview;
- physical examination;
- observation.

The interview

As part of the veterinary team, VNs regularly gain information about the *physical* status of the patient. Vital parameters are measured (temperature, respiration rate and heart rate) and organ function is evaluated through measurements of chemicals in the blood; the VN is fully aware that these values are useless unless they know what normal values should be. However does the VN gain enough information about each of their patients' distinctive and unique idiosyncrasies? Do they know what each patient's *normal* routines/behaviour/preferences are? Getting this information could make a huge difference to the approach to the VN's nursing care. For example, in the earlier example, if the VN had spoken to Dexter's owners, they might have found out that he really likes ham as a special treat, he likes to sleep on an old woollen rug with a couple of their daughter's soft toys and that he likes his chin scratched but *hates* his feet being touched.

It is frequently the veterinary surgeon who admits the patient and whilst he or she is likely to have asked the owner a number of questions about the patient during the consultation it is likely that the veterinary surgeon's questions centre around a disease-focused approach. It is therefore important that the VN can ask the owner a whole different range and style of questions in order to ascertain information that facilitates a more patient-focused, holistic approach. There may not always be the opportunity to do this during the admission of the patient (or the VN may feel that it is not the right time). If it is not done in a face-to-face meeting with the owner, the VN could telephone the owner at a later stage as a follow-up.

At whatever point the VN decides to carry out the interview, it is wise and polite to let the owner know exactly why these additional questions are being asked.

> *Mr Vann, I am Sam Smith, the qualified veterinary nurse who will be responsible for planning the nursing care Dexter will receive whilst he is hospitalised with us. I would like to spend about 20 minutes with you now, talking about Dexter's normal routine and completing his nursing admission form. This information will help me work with you to begin to plan Dexter's care.*

(In contrast to "Hi I'm Sam and I'm Dexter's nurse who will be looking after him. I just need to ask you some questions".)

A *model of care* helps to direct the questioning and help the VN systematically gain all the information from the various sources and methods listed above.

The interview (whether it be face-to-face or by telephone) requires good communication skills in asking appropriate and open-ended questions and listening will encourage the owner to provide the *relevant* information the VN needs.

Murray and Atkinson (1994) suggest that questions can be focused to guide the interviewer's questions for each area covered. The following example has been adapted from their work, to make it applicable to the veterinary scenario.

(1) What is your pet's normal pattern (or behaviour)?
(2) Has the current health problem (illness or injury) affected your pet's normal pattern (or behaviour)?
(3) Have you done anything to (or do you know of anything which might) help maintain or restore normal patterns affected by the current problem?

The following is an example of a client interview dialogue with closed questions:

VN: Do you feed Dexter cat food?
Owner: Yes.
VN: What does he eat?
Owner: Whiskas tinned food.
VN: Is his appetite good?
Owner: Yes.

From this we can ascertain that Dexter is usually fed Whiskas cat food, and that his owner thinks his appetite is good normally. Asking the questions in a different way, using Murray and Atkinson's guide for directing interview questions, could elicit more detailed information.

This is an example of a client interview using Murray and Atkinson's style of questioning:

VN: What do you normally feed Dexter?
Owner: He normally has Whiskas tinned food twice a day.

VN: Has this changed recently?
Owner: For the past three days, he's been off his food and hasn't eaten anything.
VN: Is there anything that you know of which might encourage Dexter to eat?
Owner: He goes crazy when we have ham, my 3-year-old feeds him small bits from the table.

Filling in a predesigned form during the interview can help structure and direct the questions and provide a record for use at any stage during hospitalisation. In human healthcare these forms (commonly known as nursing admission assessment forms) vary depending on the institution but most are derived from a model such as Maslow's Hierarchy of Needs or a particular nursing model such as Roper, Logan and Tierney's Activities of Living Model. These frameworks have been designed for humans so some elements may not be transferable to veterinary patients.

Physical examination

Another key element of the assessment phase is the physical examination of the patient. In the veterinary scenario, a complete clinical examination is often carried out by the veterinary surgeon at the time of admission into the practice. A complete diagnostic physical examination lies beyond the skill remit of qualified veterinary nurses; however, the VN should be competent in carrying out a systematic general physical examination, identifying what is normal and what is not. Measurement of physiological parameters at least on a daily basis is an essential part of the assessment of the patient. These measurements need to be recorded and changes referred to the veterinary surgeon in charge of the case. Further information on how this may be carried out is given in Chapter 3. The physical examination, if done skillfully and mindfully, can also be a useful way to help establish a relationship with the patient.

Observation

During the physical examination or whilst performing nursing tasks (e.g. when taking the patient out to urinate or whilst changing a bandage) the VN should consciously look at the patient, observing its body and assessing its demeanour. Does it appear depressed, agitated? Is the animal bright and alert, friendly, aggressive, or lame? The VN should use all their senses – remember, for instance, that certain diseases and conditions have characteristic odours associated with them.

Data

The data collected will be *objective* (factual), and some of it will be *subjective* (judgments or opinions from the owner). The aim is to gain *objective* data, therefore the VN should aim not to make any subjective judgments yet. For example, '*the cat looks depressed*', '*large amounts of fluid draining from wound drain*', '*dog exhausted after walk*' are subjective judgments rather than objective statements. Instead, the VN should try to be precise and describe what they have seen. For example, '*the cat is in the back of the kennel and is not responding*

to stroking or petting'; '*20 ml of pink fluid draining from wound drain*'; '*respiratory rate of 30 after walk to outside kennels and back*'.

Similarly the information gained from the interview with the owner should aim to elicit facts rather than subjective statements. For example, when asking if Dexter's appetite was good, the owner responded that generally it was. This was a judgment made by the client, led by poor questioning technique. It does not actually say how much Dexter normally eats. Asking the owner what and how much Dexter normally eats is much more likely to give a response with more objective data.

After a thorough assessment of the patient, various problems concerning the normal activities of the animal may have been identified. It is these problems that require prioritising before the goals are set.

Goals that the nurse is aiming for include:

(1) The patient gaining, maintaining or restoring maximum independence, or coping with dependence on the client.
(2) Enabling the patient/client to carry out 'preventing activities' independently to avoid ill-health.
(3) Providing comforting strategies to promote recovery and eventual independence.
(4) Providing medically prescribed treatments to overcome illness or its symptoms, leading to recovery and eventual independence.

For example, for a paraplegic Border Collie, the short-term goal may be to resume control of bladder function and be able to be supported out to the runs to urinate voluntarily. The long-term goal would be that the dog is able to walk outside unaided.

It is important that the long-term goals are identified, but in order to carry out the care plan, short-term goals must be put in place so that the care plan is achievable.

Making a nursing diagnosis

This phase of the nursing process is not always included in all nursing textbooks on the subject. However, it is an explicit element of the process; after all, one cannot plan the care if one has not decided what the care should be. It is probably helpful, therefore, at this stage to identify and define what is meant by nursing diagnosis.

The word *diagnosis* is most frequently associated with the role of the veterinary surgeon in making a *clinical* or *medical* diagnosis – by gathering information from the patient's history, clinical examination and ancillary tests, the veterinary surgeon is able to to determine and identify the underlying cause(s) of the problem or clinical symptoms.

A nursing diagnosis is not concerned with making a judgment about the disease; obviously there may be some occasional overlap, but it is really important to be absolutely clear about the demarcation. **From a legal and professional point of view, making a veterinary clinical diagnosis is beyond the remit of the veterinary nurse**.

The *nursing* diagnosis is making a judgment about the patient's health needs based on the information gained during assessment. It describes the decisions made on the care the animal requires. This may relate to its health needs but can also incorporate social and cultural issues. For example, a 40 kg Dobermann requires intensive rehabilitation after spinal surgery. The veterinary nurse has been communicating with the owner on a regular basis and is aware that the owner (an elderly frail woman) will be unable to continue some of the exercises at home. The care plan will need to take this into account.

The assessment phase helps to identify *actual* problems with the patient's health that exist at the moment and extends to identifying *potential* problems that the patient may develop. For example, Dexter has matted, faeces-covered fur around his rear end. He is not eating and his demeanour is depressed. You know that if not dealt with, these problems could lead to further potential complications. For example, he is at risk of an imbalanced fluid volume (hypovolaemia), weight loss and faecal scalding.

The NANDA (North American Nursing Diagnosis Association) has devised a set of pre-written nursing diagnoses – their rationale is that by developing a set of pre-written, accurate and valid nursing diagnoses they provide a way for nurses to analyse data and assign an accurate and valid terminology, which subsequently enables a systematic and standardised approach to patient care.

NANDA's nursing diagnoses (accessed at http://www.nanda.org/Home.aspx) provide the name of the diagnosis along with a definition which incorporates the defining characteristics and related factors. According to the NANDA system, Dexter's nursing diagnoses might be:

- bathing, hygiene and self-care deficit;
- altered faecal output (diarrhoea);
- imbalanced nutrition: less than body requirement;
- risk of deficient fluid volume.

Planning the care

The planning phase is essentially about setting goals for the patient and working out how these goals will be met by nursing interventions. From the information that has been obtained from the assessment phase, the VN will have identified a number of actual and potential nursing problems. With this nursing diagnosis, a decision can then be made on the nursing care and action required. This phase allows the nurse to set the *goals* of nursing, identify the *nursing intervention* required and, importantly, *prioritise* the order in which they should be carried out.

Set goals (outcomes)

Goal attainment is part of everyday life. Goals help us identify something that we aim to achieve so it is important for them to be realistic and achievable. In defining goals, the SMART mnemonic (specific, measureable, attain-

Table 2.2 Nursing intervention.

Problem	Goal/outcome	Time	Nursing intervention
Not eating	Dexter to eat a strip of ham	By 8 pm	Hand feed, stroke and pet Dexter. Scratch under chin

able, relevant, timely) may be used. Goals should be stated in terms of outcomes that are able to be observed, measured or tested. This allows evaluation to determine if the nursing interventions have been successful and if the goals have been achieved.

For example, Dexter has a nursing diagnosis of *imbalanced nutrition: less than body requirements* and a *bathing and self-care deficit*. The goals might be to get him eating and to get him clean and how this will be achieved will be described by the *nursing intervention*.

When setting goals, it is important to consider any other influencing factors which might have an impact on the goals that are set. For example, any cost restrictions set by the client or practice; availability of other personnel or equipment; specific instructions from the owner or vet; cultural or ethical viewpoints of the owners.

Identify the nursing intervention

Nursing interventions should be written in such a way that enables anyone involved in the veterinary care to understand what to do (Table 2.2). This means specifying exactly what is to be done, how often it is to be done, how much and in what order in relation to other nursing activities.

Prioritise

It is necessary to identify which problems need dealing with in the first instance and which are not so urgent. Clearly life-saving treatment will need to be carried out before other procedures. For example, for the RTA patient, it might be necessary to administer oxygen before cleaning the large, open wound. Preventing high-risk *potential* problems from occurring might be another factor in deciding what is done and when. For example, deciding not to groom the long-hair cat that is in heart failure.

During the second day of hospitalisation, Dexter is radiographed and a diagnosis of intussusception is made, and he requires surgery to reduce the intussusception.

Dexter's problems are:

(1) Bathing, hygiene and self-care deficit.
(2) Diarrhoea.
(3) Imbalanced nutrition: less than body requirement.
(4) Risk of deficient fluid volume.
(5) Risk of peritonitis (new nursing diagnosis since veterinary diagnosis made).

Problem 4 needs to be addressed immediately. The intervention might be veterinary-initiated. In other words, the veterinary surgeon might instruct bloods to be taken and electrolytes checked. Intravenous fluid therapy will be prescribed to improve Dexter's hydration status for anaesthesia and surgery.

Problem 5 also needs to be addressed quickly – as soon as possible after the patient has been stabilised with intravenous fluids. Obviously the resolution of this is based on the veterinary surgeon carrying out the surgery, but as a nurse you will be required to assess the patient appropriately to check for evidence of peritonitis.

Problem 1 may not need to be addressed just yet, but might need sorting out before the patient goes to theatre (to reduce the risk of infection).

Implementation

The implementation phase involves giving and documenting the actual nursing care activities as identified on the care plan. It is crucial to be specific in the care plan; everyone involved needs to know exactly what the nursing intervention is, how often it is required, and how much is required. The decision on the nursing intervention is guided by the goal(s) to be achieved, and ideally should be derived from a sound evidence base. Remember that unless a thorough and detailed assessment is carried out, you may not be in a position to carry out the most appropriate nursing care for each particular patient. If the assessment of Dexter had not revealed from the owner that Dexter loves ham the VN might end up offering him a whole variety of foods – as is often carried out in practice.

In the medical model, the choice of intervention usually involves focusing on the particular dysfunctional body system or part, the physiological problems, and administering some kind of medication to help resolve the problem. The focus of intervention in nursing models, however, is more likely to encompass not only the physiological activities, such as eating, drinking and eliminating, but also psychological, environmental and sociocultural activities which may affect the animal's recovery.

Evaluation

It is difficult to justify planning and provision of nursing care if the results cannot be shown to have benefited either the patient or the client in some way, so evaluation is an essential part of the successful nursing process. To evaluate effectively, reassessment takes place, although this is often carried out in a less formal way. The animal is assessed to discover which activities it may now be able to carry out (e.g. Dexter is now eating). The animal may, of course, have developed other problems it did not have before, so a thorough assessment is necessary not just focusing on previously identified problems. It is important to evaluate the nursing care delivered and how effective it has been. During this stage, the VN is hoping that the evaluation will show that all the nursing goals have been achieved and the patient is responding well to the nursing care provided. If this is not the case, asking

the following questions might help to determine the next step (adapted from Luker, 1989).

- Was the original goal appropriate or should it be reviewed?
- Has the goal been partially achieved? If no, then review the goal.
- Is there a worsening of the problem? If yes, then review the nursing interventions. Should the nursing intervention be changed or stopped?
- Is more information needed to decide the next step?
- Does the VN need additional skills to make the intervention more effective?
- Does the goal require interventions from other members of the veterinary team?

The correct use of the nursing process should therefore enable the nurse to:

- provide each patient with individual care of a high standard;
- communicate effectively with and involve both patient and their family in the nursing care;
- maintain continuity of care between members of the nursing team;
- communicate effectively with other professional colleagues;
- provide clear written records of nursing care for professional and legal purposes;
- develop effective methods of evaluating and reviewing care.

There are many advantages to performing an accurate nursing assessment of patients.

- Patient/client:
  - the patient's normal life can be taken into account when nursing care is planned;
  - the client has been actively involved in planning the care of their pet;
  - a better relationship can be built up between the client and nurse.
- Nurse:
  - increased understanding of the patient's needs;
  - focus on the holistic nursing of the patient;
  - a written assessment provides information that can be looked at objectively.

MODELS OF NURSING CARE

Implementing the ability model

There are many models of human nursing that can be used or adapted to inform the practice of veterinary nursing. Roper, Logan & Tierney's model is based on the 12 activities of living, considered to be the essentials requirements for being healthy. Orem's self-care model is based on the patient's self-care requisites, considering how much the patient can still do

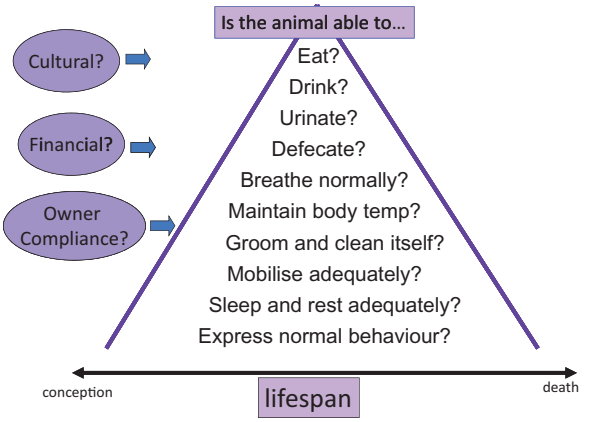

Fig. 2.2 The Orpet and Jeffery Ability model 2007.

for themselves. The ability model (Fig. 2.2) is an adaptation of both of these models; however the assessment phase focuses on the 'abilities' of the animal.

Assessment

Initial assessment of the patient can take the form of a questionnaire that the owner completes before or during the admission of their animal. The information needed for the assessment is based on the 10 'abilities' of the animal listed in Table 2.3).

It is important to remember the value of good communication skills; open questions are likely to extract more information about the patient than closed. For example; 'Does your pet eat adequately?' would probably result in a yes or no answer. However, 'What does your pet normally eat, how much and at what times?' will give a better idea. Of course finding out the animals *favourite* food is important when tempting the inappetent animal.

The next stage of the nursing assessment concerns what the animal can or cannot do now it has been admitted. Again this is done using the same checklist of 10 abilities from the model. The VN may wish to skip the questionnaire part and enter the information gained from the client directly on the assessment form. However, giving the questionnaire to the clients prior to the admission gives them an opportunity to think clearly about their answers and often clients feel that the practice values their pets and the care given to them.

The care plan

Problems will have been identified from the information gained from the assessment. It is important to consider potential problems that may occur so they can be prevented. Once the problems have been identified and prioritised, the goals and nursing interventions are decided.

Table 2.3 Assessment framework for the ability model – questions to ask the owner when admitting the animal.

Is the animal able to …	Questions to ask owner	Rationale
… eat adequate amounts?	What does the patient normally eat?	To get the patient back to eating normal food
	How much and how often?	Calculation of BER/MER may be necessary to maintain weight
	Does s/he prefer any particular type of bowl?	Persians often prefer flat bowls. Cats generally prefer china or ceramic rather than plastic
… drink adequate amounts?	How much does s/he normally drink?	The amount drunk from animal to animal will vary. It can depend on whether the animal's normal diet is wet or dry
	Do they often drink water from containers outside? Rain water?	The type of water can be important – drinking rainwater..? May prefer filtered or bottled water. Cats often drink from a tap
… urinate normally?	Where does the patient normally urinate?	Outside? Use a litter tray? Only on walks, always in the garden or on concrete?
	Does the owner use any commands?	Useful in well trained animals
	Does the patient have any problems urinating?	Does the patient have arthritis, joint stiffness? This can affect how the animal urinates
… defecate normally?	How often does the patient defecate a day?	How often the animal defecates can depend on the type of food fed
	Where is this usually done?	Some dogs have preferences on where they may urinate/defecate. Likewise cats may have a preference to the type of substrate used in their litter tray
… breathe normally?	Does the patient have any problems breathing?	This may be linked to exercise. There may be underlying disease
	Does s/he often snore when sleeping?	Facial conformation of the animal may result in breathing difficulties
… maintain body temperature?	Do you think the patient feels the cold? Does s/he have a coat in colder weather?	Very young and old animals may have problems regulating their temperature
	Where does s/he normally sleep?	Does the animal prefer warmer places to sleep or cooler places (kitchen floor etc.)?

Table 2.3 *Continued*

Is the animal able to …	Questions to ask owner	Rationale
… mobilise adequately?	Does the patient have any problems walking?	This may be answered by simply observing the animal's normal gait. It may not be possible to assess this with cats sitting in baskets
	Can s/he jump into the car easily or get upstairs?	Older animals may suffer from stiff joints and arthritis which limit the animal's normal routine
… groom and clean itself?	Does the patient normally groom itself?	Cats often spend a long time grooming and washing themselves – this is often a good sign in a hospitalised cat
	Do you groom or bath the patient normally? How often?	Longhaired animals are often groomed regularly by the owners. Find out how often
… sleep and rest adequately?	How much sleep does the patient have during a 24 hour period?	Not something the owner will usually monitor. Dogs actually spend a lot of time sleeping despite what owners think. Cats are often nocturnal, hunting at night and sleeping all day
… express normal behaviour?	Is the patient neutered? When was her last season?	Unneutered animals can display certain behaviours associated with sex hormones
	What is the patient like with strangers or other animals?	It is important to try to ascertain whether the animal displays any aggressive behaviour. Beware you may not always get accurate information on this from owners
	Does the patient have any favourite toys/chews etc.?	Toys can help settle the animal into the hospital environment
	Does the patient have any particular commands for certain activities?	Police dogs and guard dogs may have certain commands – it may be necessary to know these in advance. Many dogs have a command for urinating/defecating – this may also be useful for dogs in strange environments away from the owners
	Does the animal suffer from any sight or hearing impairment?	Often associated with old age – important to know as the animal will be unsettled even more in a strange environment

Implementing the care

Detail in the care plan is important; everyone needs to know exactly what the nursing intervention is and how often or how much is required. The decision on the nursing intervention is guided by the goal to be achieved. Setting goals is important to validate the nursing decision made and also to measure the outcomes of the nursing intervention.

Evaluation

For effective evaluation, the assessment phase should be carried out again. From each of the 10 'abilities', the VN assesses what the animal can now do or still not do by itself. Hopefully the nursing interventions have worked and the animal is now more 'able' than when admitted. If not, the VN should look again at the care given and adjust the plan as necessary.

Influencing factors

The assessment and consequent nursing care that is carried out may be affected by other factors. The life stage of the animal is an important factor to consider; neonates are unable to feed, drink, keep warm or mobilise by themselves, and this is when they are healthy. Geriatrics may have their senses affected purely by the fact they are old and mobility may be decreased due to joint pain, muscle stiffness etc.

Cultural differences may affect the care given to the animal. What role does the pet play in the owners' life – working dog? companion? breeding animal? Intensive palliative care may be required if the owner's beliefs prevent the animal from being euthanased. There may be financial implications that prevent the care that the animal requires taking place. What alternatives are there? Working with the owner, the VN may be able to teach basic care techniques or rehabilitation methods when perhaps full physiotherapy and hydrotherapy are prohibitively expensive for the client. Nursing involves not only caring for your patient but liaising with the owner regarding the nursing care given. The VN's role is invaluable in maintaining the communication between the client and the practice. VNs often speak to clients to reassure them of how their pet is progressing in the hospital. Once the animal goes home the care continues in the home. VNs need to ensure the owner is able to administer the care required, and is also compliant in doing so. Alternative solutions to a problem may be needed. It is unrealistic for an 80-year-old owner to be expected to bath their Newfoundland once a week with medicated shampoo! The influencing factors should always be considered when creating the care plan for the animal and adjustments made appropriately.

Table 2.4 is an example of an assessment of Dexter, the cat mentioned previously. From the initial assessment chart various problems have been identified, the short-term goals have been decided and the nursing interventions documented. The care plan has been drawn up (Table 2.5).

Table 2.4 Patient assessment form for the ability model.

Patient assessment form

Date of admission: 12.07.09

Date of nursing assessment: 12.07.09

Case No. 123456

Patient name: Dexter
Breed: DLH
Sex: male entire

Owner Smith
Address
Contact No.

Clinical summary (reason for admission):
Investigation into vomiting and diarrhoea, lethargy and inappettance

Owner's perception of problem:
Vomiting and diarrhoea

Previous history (surgery, disease, vaccination status, allergies)
Recently completed vaccination course (FeLV, Flu & ent.)

T 37°C

P 150

R 20

MM pale pink

CRT<2 s

Wt. 2.8 kg

Current medication
None yet – to be prescribed by VS
Analgesia PRN

Life stage:

Age: 4mths **neonate** **adult** **geriatric**

Assessment of activities of living

Ability	Usual routine	Actual problem	Potential problem
1. **eat adequate amount**	Eats ½ sachet 3x daily	Not eating, vomiting	Weight loss
2. **drink adequate amounts**	Approx. 100ml/day	Not drinking	Dehydration
3. **urinate normally**	Outside	Reduced	
4. **defecate normally**	Outside	Diarrhoea – increased water loss	Dehydration
5. **breath normally**	Normal for conformation	Ensure no difficulties arise	
6. **maintain body temp**	No problems – sleeps on boiler	None	
7. **groom itself**	Owner grooms weekly	Poor coat condition	Matted coat
8. **mobilise adequately**	No problems	Weakness and confined to kennel	Pressure sores, muscle weakness
9. **sleep/rest**	No problems – indoors mostly		Disturbed from hospitalisation
10. **express normal behaviour**	Normally active, playful	Unwell so not wanting to play	Boredom

Table 2.5 Care plan for Dexter.

CARE PLAN					
Patient name: Dexter			**Date:**		
Date	**Problem**	**Short-term goal**	**Nursing intervention**	**Reassess/ evaluation**	**Review time/date**
12th July	Not eating	To eat ½ tin of food per day	Tempt to eat by hand feeding, warming food	Has eaten ¼ tin of food	13th July
12th July	Not drinking	Maintain fluid intake at 2x maintenance requirements	Assess hydration status and monitor IVFT rate	Hydration status maintained	
12th July	Long hair unable to groom	Maintain coat condition	Groom with cat comb x2 daily Wipe any discharges from face	No matts develop and coat condition maintained	
12th July	Normal behaviour affected by condition	Encourage normal behaviour as far as possible	Regular contact with nursing staff not just when feeding or medicating. Assess pain relief requirements		

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Chapter 3

Physical Assessment of the Patient

The assessment of the patient involves not only communicating with the owner regarding the animal's needs but also measuring physiological parameters. This chapter describes the method of carrying out these basic measurements and also the structure of a basic physical examination. The VN must be able to carry out a basic physical examination and be able to recognise normal and abnormal clinical signs.

PHYSICAL EXAMINATION

On admission and at least once a day, the VN should carry out a physical examination of each patient. Abnormalities can then be identified early and treated appropriately. Temperature, pulse and respiration monitoring should be standard procedure for every animal. Further information regarding appetite, urination, defecation, vomiting, diarrhoea and any other abnormalities should be observed and recorded on the care plan. Animals recovering from anaesthesia should have particular attention paid to the monitoring of vital signs until they are fully recovered.

The physical examination of the patient provides essential information about the state of the animal's health. A routine, systematic method of examination is required to detect abnormalities and this involves practice and thoroughness. All findings must be recorded, normal as well as abnormal.

Visual inspection and observation

Procedure

(1) Observe the patient from a distance. Apprehension, fear or excitement is normal in a strange environment. However, look to see if the patient appears depressed, lethargic or distressed.
(2) Look at the overall condition of the coat and note any hair loss, scaling, pustules, injuries, parasites and wounds.
(3) Check whether the animal appears emaciated or obese and observe for any apparent lameness, weakness or neurological defects.
(4) Before handling the patient, watch the respiration rate and depth.
(5) Approach the animal for a closer examination. Do this slowly and talk to the patient whilst doing so.

Table 3.1 Normal ranges for rectal temperature.

Species	Temperature in °C	Temperature in °F
Dog	38.3–38.7	100.9–101.7
Cat	38.0–38.5	100.4–101.6
Neonatal dogs and cats	35.5–36.1	96.0–97.0
Rabbit	37.0–39.4	99.0–103.0
Guinea pig	39.0–40.0	102.2–104.2
Hamster	36.0–38.0	98.0–101.0
Gerbil	38.0–39.0	100.4–102.2
Ferret	37.8–40.0	100.4–104.2
Chinchilla	38.0–39.0	100.4–102.2
Rat	37.5–38.0	99.8–100.5

Gaining the animal's confidence will enable an easier and more accurate evaluation.

Measurement of vital signs – temperature, pulse and respiration

Taking a rectal temperature

A rectal temperature should be obtained. This is sometimes best left until the end of the examination so as not to stress the animal too much. See Table 3.1 for normal ranges.

There are two types of thermometer.

- Mercury – graduated glass bulb containing mercury. The mercury expands when it gets warm, moving along the glass tube. The thermometer may need to be shaken to ensure the mercury returns to the bulb before use.
- Digital – measures temperature electronically.

Thermometers may be calibrated in both Celsius and Fahrenheit. Celsius is now the standard unit for measurement of temperature. The conversion formula is as follows:

$$C = (F - 32) \times \frac{5}{9}$$

Procedure

(1) Restrain patient adequately.
(2) Lubricate thermometer with Vaseline, KY jelly or similar.
(3) Insert thermometer gently into the rectum and hold thermometer tip against rectal wall.
(4) Time for at least 30 seconds.

Table 3.2 Normal pulse rates.

Species	Normal heart rate per minute
Dog	60–120
Neonatal dogs and cats	200–220
Cat	100–140
Rabbit	205–235
Guinea pig	130–190
Hamster	300–600
Gerbil	100–150
Ferret	300–400
Chinchilla	100–150
Rat	260–340

(5) Read and record temperature.
(6) Clean thermometer: wipe excess faecal matter, clean and disinfect.

Recording the pulse rate

A pulse can be felt by light palpation of an artery. Each pulsation corresponds to the contraction of the left ventricle of the heart and is usually assessed at the same time as listening to the heart. See Table 3.2 for normal ranges. The pulse rate may increase with each inspiration and decrease on expiration. This is called sinus arrhythmia and is considered to be a normal variation.

A pulse may be felt at various sites where the artery runs close to the body surface:

- femoral: inner thigh;
- digital: palmar aspect of carpus;
- tarsal: medial surface of tarsus;
- coccygeal: ventral aspect of tail base;
- sublingual: under the tongue (in anaesthetised patients).

Procedure

(1) Restrain the animal.
(2) Locate pulse.
(3) Count pulsations over 1 minute.
(4) Record the rate and report abnormalities.
(5) Check the pulse rate with the heart rate to check for pulse deficits.

Recording respiration rate

Ideally respiration should be noted with the animal at rest before exciting it with an examination. See Table 3.3 for normal ranges.

Table 3.3 Normal respiration rates.

Species	Normal respiration rate per minute
Dog	15–30
Cat	20–30
Neonatal dogs and cats	15–35
Rabbit	38–65
Guinea pig	90–150
Hamster	33–127
Gerbil	40–80
Ferret	30–40
Chinchilla	40–80
Rat	70–150

Procedure

- Gently place the hands either side of the chest cavity and count the number of respirations by feeling the movement of the chest or, carefully observe the movement of the chest wall while the animal is resting, counting the number of respirations over 1 minute.
- The depth should also be noted.

Physical examination

Head

Examine the eyes, ears, nose and mouth for evidence of discharge and changes in size and shape.

Eyes

Inspect the eye and external orbital structures. The eyelids should touch the globe, but the eyelashes should not touch the surface of the cornea. Examine the conjunctiva and sclera for evidence of infection, exudates and petechiae and check their colour. Check the pupillary and consensual light reflexes.

Mouth

Gently retract the lips and examine the teeth and gums. Smell the breath, and take note of any discharges or excess saliva. Examine the mucous membranes. The capillary refill time (CRT) should be less than 2 seconds. Slow CRT indicates that peripheral perfusion is poor; this may be due to dehydration or shock. The colour of the mucous membranes should be pink. Bright red membranes may indicate septic shock or carbon monoxide poisoning, pale membranes may indicate anaemia or hypovolaemia and blue membranes may indicate inadequate oxygenation.

Table 3.4 Dental formulae for dog and cat.

	Incisors	Canines	Premolars	Molars
Deciduous dentition in the dog	3/3	1/1	3/3	
Permanent dentition in the dog	3/3	1/1	4/4	2/3
Deciduous dentition in the cat	3/3	1/1	3/2	
Permanent dentition in the cat	3/3	1/1	3/2	1/1

Examine the teeth for staining, faulty enamel, calculus, caries and loose or broken teeth. Check the dentition and note whether permanent or deciduous teeth are present. Dental formulae for dogs and cats are shown in Table 3.4.

Lymph nodes

The palpable superficial lymph nodes are (Fig. 3.1):

- submandibular;
- prescapular;
- axillary;
- popliteal;
- inguinal.

These lymph nodes should be palpated to identify enlargement indicating inflammatory, infectious or neoplastic conditions.

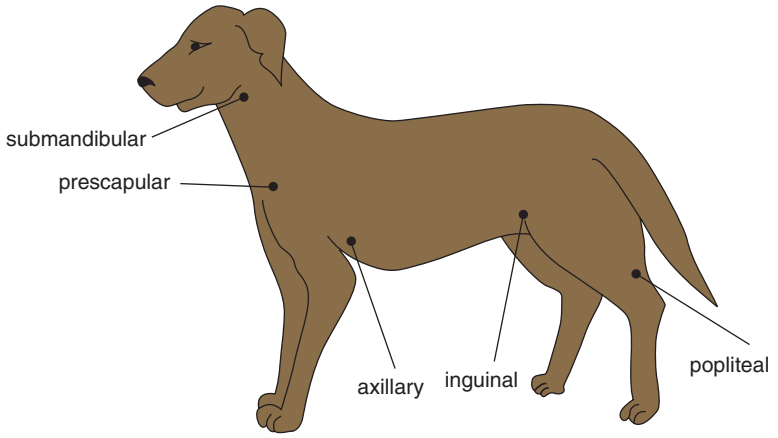

Fig. 3.1 Superficial lymph nodes.

Thorax

Auscultate the thorax using a stethoscope to assess the presence of pulmonary or cardiac problems. Listen to the heart first. Make a note of the rate and

rhythm of the heartbeat. Listen for murmurs or abnormal sounds. The normal heart sound is described to sound like 'lub-dub'. Inform the veterinary surgeon if the heart sounds are muffled. This may indicate pleural effusion, intrathoracic masses or diaphragmatic hernias. Take a femoral pulse whilst listening to the heart to confirm synchronicity. If the heart rate is higher than the pulse rate it indicates that a pulse deficit exists. Then listen to the lungs by moving the stethoscope from the dorsal thorax down to the ventral thorax. Usually soft blowing sounds are heard. Abnormal sounds such as crackles, wheezes, pops or squeaks should be noted.

Abdomen

Abdominal palpation requires skill and practice. It can potentially cause harm if carried out by the inexperienced. As a guide, the animal should be positioned on the examination table, with its head facing away from you. Without touching the animal, inspect the abdomen for general contour, presence of swelling and generalised distension. Observe the movement of the abdominal walls during respiration. Check that the animal is standing freely rather than 'tucked-up' or hunched.

Genitalia and perineal region

In the male, examine the prepuce and anus for evidence of discharge or haemorrhage. Palpate the testes to ensure symmetry. Palpate and examine the vulva and mammary glands in the female. In cases of paralysis, the anal sphincter reflex can be tested.

Limbs

Examine the lymph nodes surrounding the fore and hind limbs. Inspect and palpate the legs and joints, looking for pain, heat, swelling, deformities and restricted movement.

Weight

An accurate bodyweight for each animal should be obtained every day. Also visually assess to detect overall changes – emaciation or obesity. Losses of more than 10% of initial weight may require supportive nutrition and fluid therapy (e.g. 10 kg dog losing 1 kg or 20 kg dog losing 2 kg of weight).

Chapter 4

Nursing Activities of the Ability Model

In this chapter the framework of assessment for the ability model will be used to discuss associated nursing interventions and, in some cases, veterinary prescribed interventions.

THE ABILITY MODEL OF CARE

The assessment framework is as follows.

(1) Is the animal able to eat adequately?
(2) Is the animal able to drink adequately?
(3) Is the animal able to urinate normally?
(4) Is the animal able to defecate normally?
(5) Is the animal able to breathe normally?
(6) Is the animal able to maintain body temperature?
(7) Is the animal able to groom and clean itself?
(8) Is the animal able to mobilise adequately
(9) Is the animal able to sleep and rest?
(10) Is the animal able to express normal behaviour?

The above assessment can be used to ascertain what is normal so that the nursing care provided can help restore the animal back to normal. It is essential that the individual animal is cared for in addition to considering species-specific requirements. Registered Veterinary Nurses have agreed that by becoming registered, they will abide by the RCVS VN Guide to Professional Conduct. The first two guiding principles are perhaps the most appropriate to nursing care.

(1) *Make animal welfare your first consideration in seeking to provide the most appropriate attention for animals committed to your care.*
(2) *Ensure that all animals you care for are treated humanely and with respect.*

RCVS Guide to Professional Conduct

Five freedoms

From a welfare point of view, VNs should be ensuring both physical and mental welfare of any animal in their care. This means ensuring that the most basic requirements and needs of our patients are met.

The following is from the Farm Animal Welfare Council (FAWC) and was devised originally to ensure farm animals have adequate care. This is a useful framework that can be used for any animal.

(1) **Freedom from hunger and thirst –** access to fresh water and a nutritional diet.
(2) **Freedom from discomfort –** ensuring an appropriate environment including shelter and a comfortable resting area.
(3) **Freedom from pain, injury or disease –** by prevention or rapid diagnosis and treatment (including appropriate assessment for analgesic requirements).
(4) **Freedom to express normal behaviour –** providing adequate space, environmental enrichment and company of the animal's own kind. This may also include human company.
(5) **Freedom from fear and distress –** by ensuring conditions and treatment which avoid mental and physical suffering.

IS THE ANIMAL ABLE TO EAT ADEQUATELY?

The VN needs to have knowledge of the nutritional requirements of a wide range of species that may be hospitalised in the practice. A good diet plays an important part in the recovery and rehabilitation of a patient.

Nutrition

Nutrition is the process by which food is assimilated into the body in order to nourish it and support and maintain cell metabolism. There are six major classes of nutrients found in food.

(1) Water.
(2) Carbohydrate.
(3) Fat.
(4) Protein.
(5) Minerals.
(6) Vitamins.

These are required to produce energy for many body functions:

- growth;
- movement;
- metabolism;
- temperature regulation;
- repair of tissues;
- reproduction.

A diet containing a balance of the main classes of food will ensure there are no excesses or deficiencies of certain classes which could lead to further problems. It is important to remember that animals may require a different balance of nutrients at different life stages.

Calculation of nutritional energy requirement

Hospitalised animals may become malnourished because they are not receiving adequate calories. They are often fed single nutrient foodstuffs (chicken, fish, etc.) in an attempt to encourage them to eat. In the long term, this leads to an imbalanced diet. The hypermetabolism, that may be occurring as a result of disease, changes the normal energy requirements. A healthy animal usually loses fat as a result of starvation. A diseased or traumatised animal will often start to catabolise lean muscle mass as a result of stress starvation.

Although weighing the patient every day is an essential part of the care plan, it is not enough by itself to monitor nutritional status. Weight loss can be associated with fluid loss. Evaluating overall body condition is more useful in detecting malnourishment. There are several scoring charts available and one should be present in the ward area for reference. It is important that there is consistency in the practice as to which system is used. All hospitalised patients should have a nutrition plan and this may be incorporated in to the care plan. Even animals which do not require nutritional support should have their food requirements calculated. This can be important for the overweight animal that may require readjustment of their diet and perhaps education of the client!

There is limited research on the nutrition of clinical cases and accurate calculation of calorie requirements is debatable. Recent literature suggests creating a nutritional plan based on meeting the animal's resting energy requirements (RER). RER is defined as the number of calories required for maintaining homeostasis in a thermoneutral environment whilst at rest.

The formula for calculating RER is (BW stands for bodyweight):

$$\text{RER (kcal)} = 70(\text{BW in kg})^{0.75}$$

Alternatively, for animals weighing between 2 and 45 kg the following may be used:

$$\text{RER (kcal)} = (30 \times \text{BW}) + 70$$

This will give a reasonable estimation of the calorie requirements.

'Illness factors' have been included in the past, suggesting certain diseases may require increased calorie requirement. The work in this area has been extrapolated from studies in human medicine and it is now thought that this may result in overfeeding in animal patients, resulting in metabolic and gastrointestinal complications, and also hepatic dysfunction.

Continuous monitoring of weight and body condition will influence any changes to the nutritional plan resulting in an increase or decrease in calories provided.

Provision of nutritional support

The VN plays an important role in feeding of patients; firstly he or she needs to be aware of when certain patients may require encouragement to feed or

need to be tube fed, and report to the veterinary surgeon in charge of the case. The VN should then be part of the decision-making process to decide on an appropriate feeding regime to suit the needs of each patient's temperament and physical condition.

Part of the nursing assessment of the patient involves finding out what the animal's normal routine is with regard to feeding. This will often highlight why the animal is not eating. The type of bowl from which the animal is fed may affect feeding. Cats often prefer china flat bowls rather than plastic or high-sided bowls, especially if they have a flatter facial conformity. Behaviourists often report that cats do not naturally drink where they eat. This can be a problem in small kennels used to house cats. As previously mentioned, finding out what the animal's favourite food is should be part of the admission assessment rather than waiting until 3 days later when the cat or dog is not eating. The VN should recognise signs of nausea (hypersalivation, rejecting food, frequent licking of lips, etc.). Anti-emetics will usually be necessary before attempting to encourage the animal to eat.

Malnutrition can lead to several complications including decreased healing, decreased immune function, and increase in gastrointestinal permeability and subsequently an increase in morbidity and mortality. Signs of malnutrition include weight loss, loss of body condition, poor coat, muscle wasting and a reduction in wound healing. The main aims for nutritional support are to restore adequate hydration, correct electrolyte imbalances, maintain bodyweight and try to prevent further complications occurring as a result of the period of malnutrition.

The VN's role is to ensure the nutritional plan is carried out and continually assess the effectiveness of the plan. Nursing the patient will also involve maintenance of the feeding equipment and tubes, and ensuring accurate records of consumption are kept.

The following patients require assisted feeding.

- Anorexic animals (of more than 3 days).
- More than 10% bodyweight loss.
- Physical limitations, e.g. fractured jaw, oral ulceration, facial trauma, etc.
- Following oral surgery – too painful and gives tissue time to heal.
- Generalised loss of muscle mass.
- Generalised lethargy of more than 3 days in severely ill animals.
- Cases involving megaoesophagus.
- Conditions associated with inadequate food intake lasting longer than 3 days. It is important to get an accurate history from the owner as the inappetance will often have started before admittance and so day 1 of hospitalisation may be day 4 of inappetance.

Methods of feeding

The VN needs to assess the patient to determine what type of feeding is required. Often spending time with an animal, gently encouraging it to eat, will be enough for the animal to gradually start eating again. There are different ways in which feeding can be done.

Assisted and syringe feeding

- Encourage, talk to and stroke the animal.
- Warm moist and liquid food to increase the smell and palatability. Take care not to overheat as this will destroy the nutritional value of the food.
- Tempt the animal by placing small amounts on its lips, nose, paws, or your fingers. Some patients may not be familiar with taking food from the hand – put a small amount on the bed in front of the animal.
- Clean their face, especially nose and mouth area to enable the animal to smell and breathe whilst eating. This is especially important to cats.
- Offer small amounts and one type of food at a time (especially cats). Do not fill the kennel with bowls of food. Do not leave the food with the patient for too long – it will become cold and unacceptable.
- Use highly palatable food appropriate for each species, e.g. try oily fish for cats, adding gravy for dogs and soft, sweet fruit and vegetables for rabbits and guinea pigs.
- Fill a small syringe up with liquid food, push the tip through the side of the mouth in between the molars and empty the syringe slowly. Ensure the animal swallows before introducing more food. This can be very stressful for some patients and should only be used initially to start the animal eating.

Appetite stimulants

Various drugs act as appetite stimulants, often as a side effect of their normal use. Diazepam (Valium) may be used in cats as an appetite stimulant. This has an immediate effect so make sure the food is already prepared and ready to give as soon as the drug has been administered. Unfortunately sedation also occurs and this may not be desirable. Cyproheptadine (Periactin™), known for its antihistamine effects, also stimulates the appetite of cats.

Tube feeding

Critically ill animals or animals physically unable to eat may require tube feeding. Calculated amounts of food and water are administered via a feeding tube, which ensures that the animal receives adequate nutrition. See Table 4.1.

Intravenous feeding (parenteral nutrition)

Nutrition may be provided by the intravenous route. This is used for patients with gastrointestinal failure (e.g. inflammatory bowel disease, pancreatitis and peritonitis). Special liquids suitable for intravenous administration are used. There is a high risk of infection using this method, so solutions must be mixed aseptically and 24-hour nursing provided.

What to feed

Depending on whether the food is to be tube fed or not determines the type of food to be used. The more energy dense the diet is, the less needs to be fed. The diets in Table 4.2 are often used in assisted and tube feeding.

Table 4.1 Comparison of types of tube feeding.

Tube	Advantages	Disadvantages
Orogastric (stomach tube)	Useful in neonates and exotics	Not well tolerated by adults. Short-term use only
Oesophagostomy tube The tube is placed into the oesophagus	Useful for longer term use in cats Wider than N-O tubes	Needs general anaesthesia for placement. If animal vomits tube can be dislodged
Naso-oesophageal tube The tube is placed via a nostril into the oesophagus (size 3.5–8 Fr)	Easy to place Appropriate for up to a week Animal is still able to eat and drink with tube in place	Small-gauge tube so only liquid feeds can be used
Pharyngostomy tube The tube is placed in the lateral aspect of the pharynx into the oesophagus (size 8–18 Fr)	Useful for short to mid-term use Easy to administer any liquidised food	Requires general anaesthesia. Can be easily dislodged. Stoma site can become infected
Gastrostomy tube Placed either endoscopically or via gastrostomy (size 14–20 Fr)	Easy to feed Any liquidised food may be fed Can be left in for months at a time	Requires general anaesthesia. Specialised equipment required (endoscope)

Table 4.2 Energy density of diets for assisted and tube feeding.

Food	Energy density
Hills a/d	180 kcal/156 g can
Reanimyl	0.9 kcal/ml
Waltham Canine Conc. Instant	1.52 kcal/ml
Waltham Feline Conc. Instant	1.24 kcal/ml
Waltham Canine Conc.	137 kcal/100 g
Sensitivity Control (feline)	115 kcal/100 g

How much to feed

Once the type of food to feed has been chosen and the energy requirements of the animal have been calculated it is necessary to calculate how much food the animal requires over the day.

Divide the daily energy requirement by the energy density of the food, which should be stated on the package or tin, to find out the total volume in millilitres per day. If feeding tinned food, it is still possible to work out the required volume by knowing how many kcal/g the food contains.

$$\frac{\text{Daily energy requirement (kcal/day)}}{\text{Energy density (kcal/ml) of the food}} = \text{amount (in ml) to feed}$$

Example:

An animal requires 1050 kcal per day and the energy density of the chosen food is 1.2 kcal/ml.
1050 kcal ÷ 1.2 kcal/ml = 875 ml per day.
This amount may then be divided up into smaller meals to be fed throughout the day.

Continuous feeding

Continuous feeding may be administered via drip bag or syringe pump at a rate of 1 ml/kg/hour. This method is often used with jejunostomy tubes. The rate is gradually increased until the daily volume can be fed over 12–18 hours.

Repeated bolus

Giving a repeated bolus is perhaps a more natural way to feed but can depend on whether the patient can tolerate this. A calculated quantity is given divided throughout the day. Up to 30–45 ml/kg is given per feed. The stomach capacity is approximately 50–90 ml/kg, but this is reduced following periods of inappetance.

How to feed using a naso-oesophageal tube

(1) Place a few drops of local anaesthetic around the tube into the nostril about 5 minutes before the feed. This prevents the animal being irritated by any movement of the tube as it is fed.
(2) Warm the food to body temperature.
(3) If it is not certain whether the tube is in place, flush first with 1–2 ml of sterile saline or radiograph the patient.
(4) Flush the tube with 5–10 ml of water (warm).
(5) Slowly administer the required amount of food. Use 20 ml syringes as less force is required to syringe the food.
(6) Observe the animal, ensuring it is showing no signs of discomfort.
(7) After administering the food flush the tube again with water to clear the tube of food.
(8) Increase the amount gradually starting with one third of the daily allowance over the first day.
(9) For the first 24 hours, dilute the food by 50% to reduce the chance of diarrhoea.

How to tube feed using an oesophagostomy tube

The increase in tube diameter compared to the naso-oesophageal tube means that diets of slightly thicker consistency may be used. The same precautions apply for preventing blockage. The food should be fed slowly over 5–10 minutes (Fig. 4.1). Dislodgement of the tube can occur if the animal vomits. Care of the stoma site is also important to prevent cellulitis and infection.

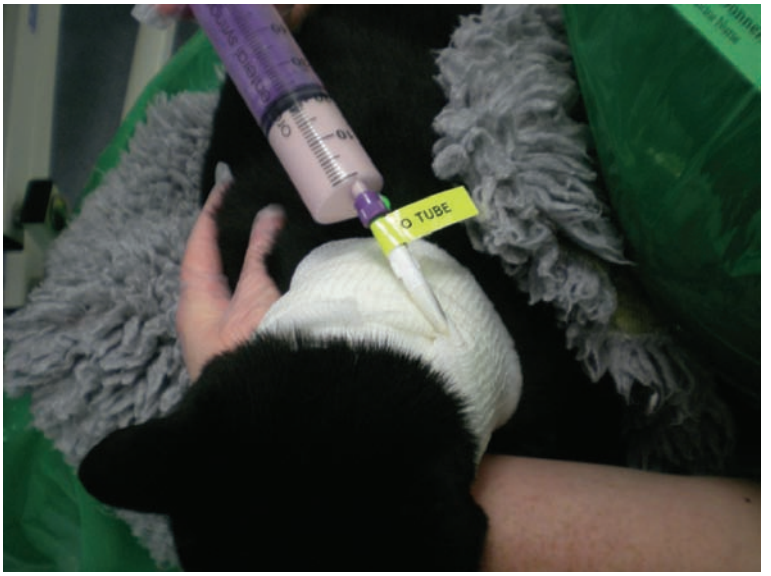

Fig. 4.1 Oesophageal tube feeding.

How to feed using a gastrostomy tube

(1) Do not use for first 24 hours, allowing time for a primary seal to form between the stomach and body wall.
(2) Start with small amounts of water (5 ml/kg) to flush the tube.
(3) At first, feed only a third of the daily requirements divided into several small meals. Increase to normal amount over 3 days.
(4) Ensure the food is warmed to body temperature and administer slowly, observing the animal for any signs of discomfort (Fig. 4.2).
(5) Always flush the tube after feeding.
(6) Check placement of tube and clean wound if necessary.
(7) Replace the bandage over the feeding tube.

Care of feeding tubes

- Radiograph to ensure correct positioning of distal end of tube (most naso-oesophageal tubes have a radio-opaque line).
- Test the tube each feed by syringing 1–2 ml of sterile saline (if it goes into the trachea, the saline will be absorbed quickly). If the animal coughs, check the position as shown above.
- Always flush tubes after feeding to prevent clogging of tube with food.
- Replace spigot after use.
- Unclog blocked tubes by flushing with water or fizzy drink – the bubbles will help break down the blockage.
- Check tube entry site daily and cover with dressing and bandage if necessary (gastrostomy tubes and oesophagostomy).

Fig. 4.2 Gastrostomy tube feeding.

- Prevent patient interference with Elizabethan collar or bandage.
- Check patient for evidence of vomiting, regurgitation or bloating.
- Check for diarrhoea.
- Check bodyweight daily.
- In the event of aspiration of food, stop feeding, report to the VETERINARY SURGEON and remove the tube.

Care of the patient with feeding tubes

In addition to monitoring vital signs, it is beneficial to palpate the lymph nodes nearest the tube insertion for evidence of enlargement, indicating infection at the tube site.

When oesophagostomy or gastrostomy tubes have been placed, the following action should be taken.

- Cover the tube insertion area with povidone–iodine ointment and a sterile keyhole dressing.
- Cover entire area with padding and bandage to prevent the tube dangling or becoming dislodged. Stockinet bandage is particularly useful as a final layer over the padding.
- Clean the incision site daily with antiseptic soap and water.
- Flush the tube with 5 ml of water before and after feeding to prevent tube blockage.
- Always ensure that the end of the tube is plugged before and after feeding (an intravenous needle cap may be used if a spigot is unavailable).
- Prevent patient interference with an Elizabethan collar.

- Observe the patient for evidence of vomiting, regurgitation or bloating.
- Check for diarrhoea. If present, you may need to dilute feeding solution.
- Monitor bodyweight daily.
- In the event of tube displacement or aspiration of food, stop feeding and remove the tube.

When a nasogastric tube has been placed, the follow procedures should be followed.

- Ensure that the tube is still secured to the animal's facial skin.
- Check the tube frequently to ensure that the distal end has not become displaced (observe for coughing).
- Ensure feeding solution is sufficiently dilute for the liquid to pass through the narrow bore of the tube.
- Test the tube position before each feed by syringing 1–2 ml of sterile saline (saline will be absorbed quickly if inserted into the trachea).

IS THE ANIMAL ABLE TO DRINK ADEQUATELY?

Monitoring fluid intake is essential in the hospitalised patient. Inappetance or inability to eat or drink will lead quickly to dehydration. Water accounts for approximately 60% of the animal's bodyweight (Fig. 4.3). In younger animals this can be up to 70%. Obese animals' fluid requirements should be based on their ideal weight as fat contains much less water than other organs or tissues.

There is a continuous exchange of water and electrolytes between the fluid compartments of the body. A loss from one compartment (e.g. haemorrhage – loss from intravascular compartment) will cause losses from the other compartments as the body attempts to compensate. With loss of fluid, other substances such as electrolytes are also lost.

- The main electrolytes in intracellular fluid are potassium and magnesium.
- The main electrolytes in extracellular fluid are sodium and chloride.

These electrolytes are essential for normal function of the body.

To maintain fluid balance in the body, intake must equal output. Intake of water comes from drinking, eating and a small amount from metabolic activity. Loss of water from the body is from urination, defecation, respiration and sweating. Additional losses occur in the ill animal from vomiting, diarrhoea, haemorrhage and discharges. To maintain normal fluid balance in the body a volume of 40–60 ml/kg over 24 hours is required. In illness and increased loss this is obviously higher.

Encouraging the animal to drink

Most pets are given water from the tap and freely drink this. Sometimes the amount of dissolved minerals in the water is unpalatable to the animal. Many cats will prefer to drink rain water from buckets outside rather than fresh tap

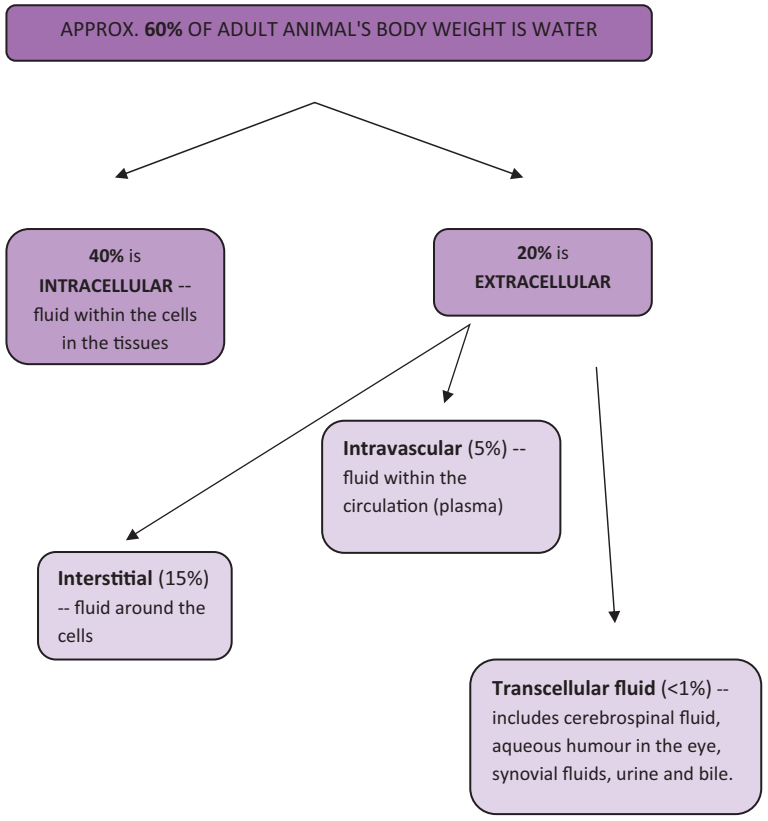

Fig. 4.3 Distribution of water.

water. More recently owners have reported cats preferring to drink from running water. A number of commercially made pet fountains are now available providing the animal with constant running water. Owners may also report their pet prefers bottled water, again most likely due to the absence of minerals found in tap water. This sort of information can be obtained at the initial nursing assessment. Getting the animal to eat wet food or adding water to the food may help to ensure the animal has some fluid intake.

Monitoring fluid intake

If the animal is drinking unaided, it is easy to calculate the total quantity of fluids that the animal is consuming by placing a measured amount in a non-spill bowl and then measuring what is left at a later time. All details must be recorded on a chart such as the one shown in Table 4.3.

If the animal's intake does not match daily requirements, additional methods of providing rehydration will be required. (Do not forget to consider quantity of water in any food given.) As with force feeding food, repeated forced application of oral fluids is not recommended, as the animal may

Table 4.3 Fluid therapy chart.

Date	Time	Quantity given	Time checked	Quantity left	Amount consumed	Notes
1/1/09	8.00	500 ml	14.00	475 ml	25 ml	4 ml/hr
1/1/09	14.00	500 ml	24.00	300 ml	200 ml	20 ml/hr
2/1/09	00.00	500 ml	10.00	450 ml	50 ml	5 ml/hr

aspirate fluid and it is stressful. Animals which are not eating but are still drinking may be given oral rehydration products which contain water and electrolytes. Examples of oral rehydration products include Pedigree Electrolyte Instant Fluid™ and Lectade™. These are suitable only for cases of mild dehydration.

Assessing dehydration

It is essential that the VN is able to recognise the need for fluid replacement therapy. Careful monitoring of hospitalised patients ensures that this is not overlooked. If the patient has just been admitted it is important that that the nursing assessment of the animal with regards to eating and drinking gains information on the following:

- appetite (normal and recent);
- water intake (including water in food, milk etc.);
- any vomiting;
- any diarrhoea;
- reduction in urination (oliguria);
- any discharges (pyometra, wounds, blood loss, burns);
- general demeanour – lethargy or depression.

A clinical examination is essential in order to assess the vital signs and obvious clinical signs associated with dehydration:

- temperature (pyrexic);
- pulse (rapid, weak);
- respiration (panting);
- mucous membranes (dry, pale);
- capillary refill time (slow?);
- sunken eyes (the tissues lose water);
- cold extremities (poor circulation);
- loss of skin elasticity (loss of water in the tissues);
- bodyweight.

A rather rough guide to the level of dehydration is gained by assessing the clinical signs (Table 4.4). A more accurate method of assessing dehydration is by analysis of blood and urine in the laboratory (Table 4.5).

Table 4.4 Clinical signs of dehydration.

Percentage dehydration	Clinical signs
<5%	Lethargy, thirst
5–8%	Skin starts to tent, slightly sunken eyes, dry mucous membranes, slow capillary refill time (CRT)
8–12%	Marked skin tenting, CRT >2 s
>12%	Shock, death!

Table 4.5 Laboratory values to assess dehydration.

Test	Normal values		Lab results associated with dehydration
	Dogs	Cats	
Packed cell volume (PCV)	37–55%	22–45%	**Increased** – the loss of water will cause the blood to be more concentrated, however if there is a loss of blood from haemorrhage then the PCV may decrease
Total protein	6.0–7.8 g/dl	6.0–7.5 g/dl	**Increased** due to reduced plasma water
Urine specific gravity	1.015–1.045	1.015–1.060	**Increased** due to reduced water content of urine
Urine output	20–30 ml/kg/day	20–30 ml/kg/day	**Decreased production –** urine becomes more concentrated Output <1–2 ml/kg/hour
Central venous pressure	3–7 cmH_20	3–7 cmH_20	**Decreased –** low circulating volume

Packed cell volume. If the normal is not known for that particular animal an estimate of 45% for dogs and 35% for cats is made. A 1% increase in PCV is equal to approximately a loss of 10 ml/kg bodyweight. For example, a PCV of 55% in a 20 kg dog would represent a loss of 2000 ml:
20 kg × 10 ml/kg × (55 – 45) = 2000 ml.

INTRAVENOUS FLUID THERAPY

The VN should be competent in placing peripheral intravenous catheters. The placement of central lines may also be carried out by experienced nurses. This is covered in Section 8.

Fluid therapy solutions

There is a tendency to use the same solution for fluid therapy no matter what the cause. Generally this is not a problem in the short term as the main goal is to correct the fluid deficit. Hartmann's is a good choice for most conditions

as it contains electrolytes in similar concentrations to those in extracellular fluid. It is also slightly hypotonic allowing it to enter the tissue cells easily. For each case, one should consider which electrolytes are being lost and replace like with like. In conditions where urinary output has failed (e.g. urethral obstruction, acute renal failure, bladder rupture), metabolites and electrolytes which are normally excreted in the urine accumulate in the body. This will also lead to an imbalance.

Crystalloid solutions

This is a group of sodium-based electrolyte fluids. Crystalloids enter the extracellular fluid and equilibrate with other fluid compartments to restore fluid balance. Most commonly used crystalloids are similar to extracellular fluid in composition.

Colloid solutions

Colloids are a group of fluids containing large molecules that will remain in the intravascular space. They help to expand the circulating volume of the blood so are also referred to as plasma expanders. They also help to increase the osmotic pressure preventing further plasma loss into cells and interstitial spaces (see Table 4.6).

Whole blood

Whole blood should be used in cases of:

- severe haemorrhage;
- severe anaemia;
- blood clotting disorders.

Blood products

- Plasma may be separated from the cellular part of blood and administered to replace plasma proteins. It also minimises the risk of transfusion reactions.
- Packed red cells may also be given to replace cell loss. They are administered simultaneously with 0.9% saline solution.

Calculating rates of administration

A fluid plan needs to be worked out in order to calculate total fluid requirements. Existing deficits, maintenance and continuing losses all need to be taken into account.

Rehydration

The amount of fluid required to correct the existing deficit may be estimated in the following ways.

Assessing from clinical signs how dehydrated the animal is

$$\%\ \text{dehydration} \times \text{bodyweight (kg)} \times 10 = \text{volume (ml) of fluid}$$

Table 4.6 Summary of fluid therapy solutions.

Crystalloid Solutions	Osmolarity	Composition	Indications
0.9% normal (physiological) saline	Isotonic	Na, Cl	Loss of chloride, e.g. vomiting and general extracellular fluid replacement
7.5% saline	Hypertonic	Na, Cl	Acts as a plasma expander by moving water into the vessels – short term only
Compound sodium lactate (Hartmann's)	Isotonic/slightly hypotonic	Na, Cl, K, lactate, Ca	Use in cases of acidosis, e.g. diarrhoea and general extracellular fluid replacement
0.18% NaCl and 4% dextrose	Isotonic	Na, Cl, dextrose	Good maintenance fluid
Ringer's solution	Isotonic	Na, Cl, K, Ca	Use in alkalosis, e.g. persistent vomiting
5% dextrose	Isotonic	Dextrose	Water losses
Colloid solutions	**Type**	**Composition**	**Indications**
Haemaccel/Gelafusin	Gelatins	Na, Cl, gelatins	Restore circulating volume
Pentastarch, Hetastarch	Starches	Starches	Restore circulating volume
Dextrans	Dextrans	Na, Cl, dextrose	Restore circulating volume
Plasma	Natural	Plasma	Restore circulating volume
Whole blood	Natural	Whole blood	Replacement of blood, plasma, platelets and colloids
Oxyglobin™	Natural	Bovine haemoglobin	Restores oxygen-carrying capacity and acts as a plasma expander

Example:

A 20 kg dog is 5% dehydrated.
$20 \times 5 \times 10 = 1000$ ml
Therefore approximately one litre is required to replace the existing deficit.

Assessing fluid loss from an increase in PCV

$$(\text{PCV measured - normal PCV}) \times \text{bodyweight} \times 10 = \text{volume}$$

Example:

A 20 kg dog has a PCV of 50%.
$(50\% - 45\%) \times 20 \times 10 = 1000$ ml

Assessing loss from history from owner

Losses from vomiting and diarrhoea etc., multiplied by the number of days.

Example:

A 20 kg dog has had no food or water for 3 days and has been vomiting three times daily for 3 days.
3 days vomiting at 4 ml/kg/vomit = 240 ml

Maintenance

Maintenance equals 50 ml/kg bodyweight/24 hours.
For example, maintenance for a 20 kg dog: 50 × 20 = 1000 ml/24 hours.

Continuing losses

As well as rehydration and maintenance fluids, compensation must also be made for continuing losses through vomiting and diarrhoea. Estimated losses (in ml) are added to the fluid plan.
Example:

A cat vomits approximately 20 ml of fluid and excretes approximately 40 ml of watery diarrhoea daily.
Therefore 60 ml of fluid needs to be replaced and must be added to the 24-hour fluid plan.

The whole fluid plan

A 40 kg dog is estimated to be 8% dehydrated. The dog is vomiting approximately 400 ml each day. What will the fluid requirements be for the first 24 hours?

Rehydration	3200 ml
Maintenance	2000 ml
Continuing loss	400 ml
Total	5600 ml/24 hours

Rate of fluid replacement

Factors that affect the rate of infusion include:

- rate of loss – e.g. acute loss;
- health of patient – e.g. elderly animal with renal insufficiency;
- type of fluid – e.g. crystalloid, colloid, whole blood;
- ongoing losses – e.g. vomiting, diarrhoea, discharges.

Blood volume in adult animal = 90 ml/kg.
Therefore, the *maximum rate* of infusion of crystalloids should not exceed 90 ml/kg/hour for 1 hour only. It is possible to administer 20–30 ml/kg/hour for less severe dehydration or administration of colloids. Normal maintenance fluid rates are 1–2 ml/kg/hr.

Working out the drip rate

Different giving sets administer different amounts of drops per ml. For example, paediatric sets (burette) deliver approximately 60 drops/ml, whereas most other sets deliver 20 drops/ml. Paediatric or burette sets are more accurate and should be used on cats and small dogs to prevent overinfusion.

Method:

(1) Calculate maintenance requirements.
(2) Divide the total by 24 to obtain amount per hour.
(3) Then divide by 60 to obtain ml per minute.
(4) Multiply this figure by the drip factor (either 20 drops per ml or 60 drops per ml).
(5) This will give the number of drops per minute.

In summary:

Total amount required in 24 hours divided by 24 = hourly rate
Hourly rate/60 = minute rate
Minute rate × administration drip factor (e.g. 20) = drops per minute

Example calculation

A 15 kg dog requires 1500 ml over 24 hours. What rate should the fluids be set at using a giving set that delivers 20 drops/ml?

Answer:

1500/24 = 62.5 ml/hour
62.5/60 = 1.04 ml/minute
1.04 × 20 = 20.8 drops/minute
The giving set should deliver 1 drop every 3 seconds.

Equipment and technique

All equipment must be prepared before placing the intravenous catheter. Pre-fill the T-connector with heparinised saline (one unit heparin per ml of 0.9% saline), cut lengths of securing tape and warm the fluids to body temperature. Fluid pumps are becoming more popular, and these provide accurate dosing of fluid. Syringe pumps are useful in small pets to deliver lower volumes.

Method of setting up the fluid bag and giving set.

(1) Check expiry date of the fluid bag.
(2) Warm the fluids to body heat*.
(3) Remove outer package.
(4) Break seal to giving port and open giving set bag and close off the giving set regulator.
(5) Insert giving set into bag (do not contaminate the set).
(6) Squeeze the drip chamber to fill half full.

(7) Open the regulator and allow fluids to fill up giving set tubing slowly. The slower the fluid fills the tubing, the less likely bubbles will appear in the tubing. If large bubbles form these should be flushed through before attaching to the catheter.

*The best method to warm fluids is in a water bath or heated cabinet. Microwaving the fluid can cause 'hotspots' within the bag. In most cases the fluid need not be warmed, as by the time the fluid has been run through the tubing to the patient it will usually have lost a significant amount of heat. Fluid warmers which include the line are more appropriate (Fig. 4.4).

The fluid supply is connectred to the catheter ensuring the distal end of the giving set remains sterile. A bandage is applied over the site to cover the catheter and giving set. The site should be examined daily for evidence of infection or swelling.

Monitor the patient.

- Check fluid is flowing at the correct rate.
- Flush line with heparinised saline if blocked.
- Do not flush with force – may need to change catheter.
- Extend animals leg – may be a positional blockage.
- Prevent patient interference.

Intravenous catheters

The main nursing consideration for patients with an intravenous catheter is to maintain a patent line and prevent sepsis. Complications are more likely

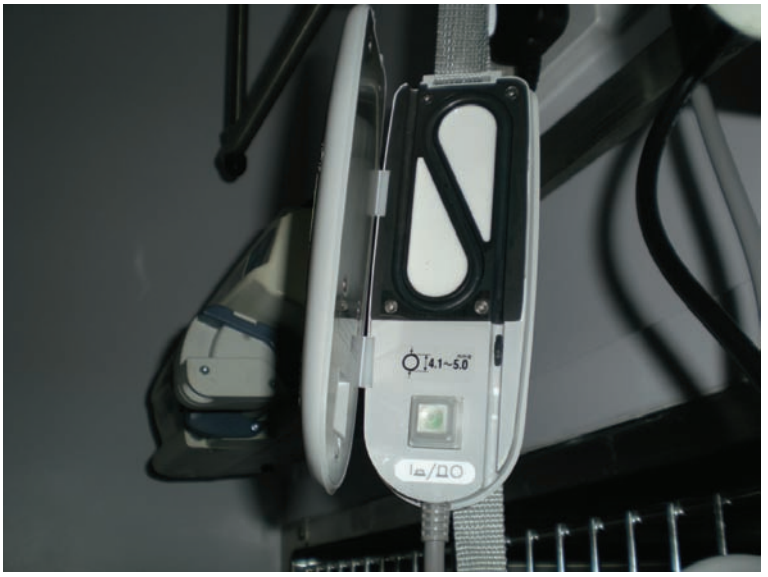

Fig. 4.4 In-line fluid warmer.

the longer the catheter is left in place, so strict aseptic techniques should be applied to prevent nosocomial infection.

- Wash hands before touching the catheter and associated connectors or fluid lines.
- Examine catheter site for signs of infection (erythema, swelling).
- Palpate lymph nodes above and around catheter site each day during placement.
- Obtain core body temperature twice daily to detect pyrexia.
- Wipe catheter junctions and bungs with 70% isopropyl alcohol before handling or injecting any drugs.
- Flush the catheter two to four times a day with heparinised saline to prevent coagulation and thrombus formation.
- Administer drugs slowly.
- Clamp off the proximal line on any administration lines to ensure that the drug enters the patient and not the fluid bag.
- Apply a topical antibiotic preparation or povidone–iodine over catheter entry site to reduce the risk of catheter-related sepsis.
- Bandage entire catheter in place to help prevent contamination and patient interference.
- Use a splint or bandage to extend the limb to prevent kinking of the catheter.
- Change bandage if it becomes soiled.
- Remove the catheter as soon as it is no longer required.
- Change catheter site every 3 days (7 days for jugular catheters).
- Take care when removing the bandage and catheter. Cut bandages on the opposite aspect of the limb from the catheter to prevent cutting through the catheter needle.

Central venous pressure

Central venous pressure (CVP) is a method of measuring venous blood pressure in the anterior vena cava (Fig. 4.5). The measurement indicates the

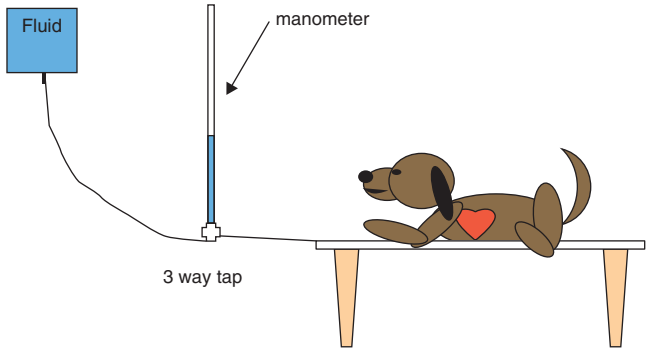

Fig. 4.5 Measurement of CVP.

heart's ability to pump venous return and the adequacy of the circulating blood volume. It aids clinical evaluation of patients with reduced blood volume and impaired cardiac function and is of particular benefit in hypovolaemic, geriatric and surgical cases. The CVP measurement is the first parameter to change when the circulation is compromised and the last parameter to return to normal during recovery and it is for this reason that it is a valuable guide to the animal's response to treatment, much more so than other physical signs such as temperature, pulse quality and mucous membrane colour.

Normal values

There is no absolute normal value for CVP and there is a wide range in the cat and dog. What is more important and of greater value, is to obtain frequent readings and thus establish a trend. The findings can be recorded and an appropriate response to increases or decreases implemented as necessary.

The normal CVP range is 0–5 cmH_2O. Increases above the normal range indicate expanded blood volume, either due to fluid replacement being administered too quickly or the heart or kidney's inability to cope with the infusion. Increases above 15 cmH_2O indicate circulatory overload and all fluid administration should be stopped. Decreases in CVP indicate that blood volume replacement is indicated or that the current fluid administration rate is inadequate.

IS THE ANIMAL ABLE TO URINATE NORMALLY?

The VN must ensure that all animals are given the chance to urinate and defecate in a way that is most comfortable for them. For example, cats need clean litter trays and some may desire more privacy (place litter tray inside a box). Some dogs will only urinate on grass whilst others will urinate only on concrete. The nursing assessment will be able to establish these facts. Animals should be taken out as often as appropriate. Polydipsic patients and animals receiving diuretic treatment will require more frequent opportunities to urinate. Some patients may be under strict veterinary instructions not to be taken outside. This may include cardiac conditions and postoperative orthopaedic cases. Unfortunately this may cause the animal stress as it is normally very well house trained. If appropriate lay some newspaper or incontinence sheets on the ground outside the animal's kennel and allow it to urinate on these.

Monitoring urine output

Observation and consequent recording of urination patterns in the hospitalised animal is an essential part of the care required. Production of urine is a good indication of kidney function. On most record systems there is only the space to tick whether it has urinated. It is also important to indicate whether any straining was present and the amount produced (normal = 1–2 ml/kg/hr). Accurate urine production requires the placement of a urinary catheter attached to a collecting bag.

Technique for manually expressing the bladder

By squeezing the bladder gently so that pressure increases, the urinary sphincter is opened and urine should flow out.

- Place a hand on either side of the caudal abdomen over the bladder.
- When you can feel the bladder, apply gentle continuous pressure.
- Diazepam and phenoxybenzamine (Dibenylin) may be administered according to the veterinary surgeon's instructions to help relax the urinary sphincter. You can then manually express 20–30 minutes later.

Urinary catheterisation

Urinary catheterisation requires passing a catheter into the bladder via the urethra. Indications are summarised in Table 4.7. There is a risk of iatrogenic trauma and infection and so the VN must ensure that the correct and aseptic technique is used.

Complications with catheterisation

Infection

Urinary tract infection (UTI) may occur easily if bacteria in the urethra are pushed into the bladder. The risk of infection is increased when:

Table 4.7 Indications for urinary catheterisation.

To obtain a urine sample	When the patient will not urinate Cystocentesis is the preferred method for bacteria culture and sensitivity tests
Monitor urine output	Check kidney function During intensive care if the patient is sedated or recumbent After renal surgery to ensure adequate production of urine (>1–2 ml/kg/hr)
Maintain patent urethra	Cats with feline urological syndrome (allows drainage whilst treatment is initiated) To relieve dysuria or anuria in the non-surgical patient
Maintain constant and controlled bladder drainage	In recumbent or comatose patients to prevent soiling After bladder surgery to prevent tension on suture line
To empty the bladder	Before abdominal, vaginal or urethral surgery Before radiography – pneumocystogram To relieve urine retention
Introduce contrast media	For radiographic procedures – bladder investigation or to check the patency of the urethra
Hydropropulsion methods	Use of water pressure to dislodge particles in the urethra
Introduction of drugs	Antibiotics etc. may be introduced although this is not common

- the tract is traumatised;
- aseptic technique is not used;
- there is presence of vaginal or preputial discharge;
- indwelling catheters are used;
- the patient is immunosuppressed and therefore susceptible to infection;
- there is repeated catheterisation.

Trauma

Causes of trauma.

- Some epithelial damage may occur in male dogs as the catheter is passed around the ischial curve.
- Force applied during catheterisation.
- Inadequate lubrication of the catheter.
- Patient resistance.
- This is an uncomfortable procedure, especially in bitches or queens and it may be necessary to sedate or anaesthetise the animal.
- Blockage. May be due to small calculi blocking the catheter – flush with sterile saline or water.

Patient interference

Patients are likely to remove indwelling catheters; an Elizabethan collar should be used and the catheter must be sutured adequately.

Equipment for catheterisation

Catheters come in a variety of shapes, sizes and materials and are designed for single use only.

Additional equipment required for catheterisation.

(1) Speculum:
- metal instrument used to enable visualisation of the urethral orifice;
- auroscopes may be adapted also for this use;
- must be sterile.

(2) Light source: pen torch/head light.

(3) Stylet:
- to aid the use of the more flexible catheters (e.g. Foley);
- usually made of metal, easily sterilised.

(4) Syringe: for inflating/deflating Foley balloon.

(5) Bungs and spigots: to block the distal free end of the catheter if left as an indwelling catheter.

(6) Three-way tap: useful when draining the bladder.

(7) Urine collection bags:
- pre-packed and sterile for single use only;
- one-way valve ensures no urine can track back;
- empty drip bags with giving sets may also be used but can be difficult to empty in a sterile manner.

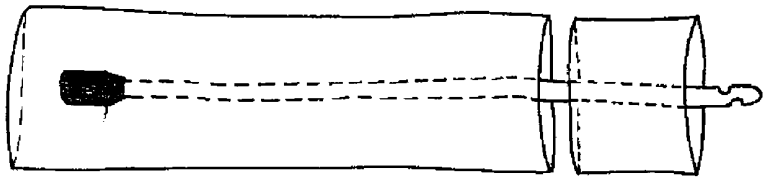

Fig. 4.6 No-touch method of handling a urinary catheter.

General points for methods of urinary catheterisation

Adequate restraint is essential, especially if performing this on the conscious patient. Bitches and queens are usually anaesthetised. Ensure all the equipment is prepared in advance. Catheters must be placed using an aseptic technique to prevent nosocomial infections occurring. Sterile surgical gloves, ideally, or disposable gloves should be worn after washing the hands. KY Jelly™ is the most commonly used lubricant, and will not damage latex. The sterile disposable sachets should be used or a new tube. Local anaesthetic, e.g. 2% lidocaine hydrochloride gel (Xylocaine™), may be used as it helps to desensitise the prepuce or vestibule. Clip and clean around external genitalia if necessary, removing discharges or dirt. Rinse the area to remove any traces of soap. Do not insert an over-long catheter too far – it can bend, kink, re-enter the bladder or damage the bladder wall. Estimate the length of the catheter to be inserted. Never twist the hard polyurethane catheters harshly whilst in the urethra as this can cause trauma and never apply digital pressure to the bladder when the catheter is in place because the catheter may traumatise the bladder.

A 'feeder' section of the catheter package can be used to ensure a no-touch method of inserting the catheter (Fig. 4.6). Once the catheter is in the bladder, urine should begin to flow into the package. Discard the first few millilitres and collect 6–10 ml in a sterile syringe if required for analysis.

Techniques

Catheterisation of male dogs

(1) An assistant restrains the dog in lateral recumbency and holds the upper portion of the hind limb away from the site of operation.
(2) Expose just the tip of the catheter and apply sterile lubricant.
(3) Retract the prepuce to expose the penis.
(4) Use the other hand to pass the catheter into the urethral opening.
(5) Continue feeding the catheter along the urethra gently (it should pass easily – if this is not the case, re-evaluate the size of the catheter).
(6) When the catheter reaches the ischial arch, gently rotate the catheter and continue to push with steady pressure.

Catheterisation of female dogs

(1) An assistant restrains the dog in standing position and holds the tail to one side, away from the site of operation (some operators prefer the dog to be in lateral recumbency).

(2) Open the inner pack and apply sterile lubricant to the tip of the catheter.
(3) Rest the catheter on the opened sterile pack.
(4) Lubricate the sterile speculum (vaginoscope, otoscope or nasal speculum can be used).
(5) Using one hand, insert the speculum into the vagina and direct the tip first dorsally, then cranially (to avoid the clitoral fossa).
(6) Visually locate the urethral orifice on the ventral floor of the vagina approximately 3–5 cm inside the vagina.
(7) Use the other hand to pass the catheter into the urethral opening.
(8) Withdraw the speculum, taking care not to dislodge the catheter tip.
(9) Continue feeding the catheter along the urethra gently (it should pass easily – if this is not the case, re-evaluate the size of the catheter or use a stylet with Foley catheter).

Some people prefer not to use a speculum when catheterising bitches. Instead they locate the urethral opening manually. This technique takes more practice initially but is less awkward than trying to manage the speculum and catheter at the same time. Using a gloved, lubricated finger, insert into the vagina dorsally and then cranially for approximately 3 cm. Gently palpate the vestibular floor to feel for the urethral papilla, which feels like a soft, round mass 0.5–1 cm in diameter, just cranial to a slight depression in the vaginal floor. Placing the finger on top of the papilla the other hand passes the catheter beneath the papilla down into the urethra. If the catheter can be felt passing directly underneath the finger, it is misdirected and should be re-positioned correctly.

Catheterisation of male cats

(1) General anaesthesia or heavy sedation is usually required for urinary catheterisation of the male cat.
(2) An assistant restrains the cat in dorsal recumbency and holds the hind limbs cranially away from the site of operation.
(3) Expose just the tip of the catheter and apply sterile lubricant.
(4) Retract the prepuce to expose the penis and gently put traction on the penis in a caudal direction to help straighten the penile flexure.
(5) Use the other hand to pass the catheter into the urethral opening (keeping the catheter parallel to the spine).
(6) Continue feeding the catheter along the urethra gently (it should pass easily – if this is not the case, re-evaluate the size of the catheter).

Catheterisation of female cats

Sedation or general anaesthesia is required to perform urinary catheterisation of the female cat. It is a difficult procedure and one that is rarely carried out.

(1) An assistant restrains the cat in lateral recumbency and holds the tail to one side away from the site of operation.
(2) Expose just the tip of the catheter and apply sterile lubricant.
(3) With one hand insert a small lubricated sterile speculum into the vagina and locate the urethral orifice on the vestibular floor.

(4) With the other hand, pass the catheter into the urethral orifice.
(5) Continue feeding the catheter gently along the urethra.

Care of patients with urinary catheters

Studies have shown that after 4 days of closed urine collection, most catheterised patients develop UTIs despite being given antibiotic therapy. It is recommended, therefore, that an indwelling urinary catheter should not be left in place for more than 24 hours and that a closed collection system is used to help prevent UTI. You must observe the patient closely for development of pyrexia, discomfort, pyuria and other signs of UTI.

- Wash hands and wear disposable gloves (for your own safety precautions) before handling and checking the catheter and lines.
- Ensure that the urine drainage bag is lower than the level of the patient's bladder to prevent urine flow back.
- Check lines and catheter to ensure they do not become kinked or blocked.
- Prevent self-trauma by applying an Elizabethan collar.
- Apply an antibacterial ointment to the urethral orifice.

IS THE ANIMAL ABLE TO DEFECATE NORMALLY?

As with urination – it is essential to observe and record the frequency and type of faeces being produced. It is important to remember what goes in must come out in some form or other! An inappetent animal is unlikely to produce a lot of faeces. A dehydrated patient may produce hard dry faeces. It is also important to note if blood is being passed or the animal has particular difficulties passing faeces. From the nursing assessment the normal routine of the animal will be obtained, however with the change of environment and probably change of diet, this may become irregular. Ensuring the animal has the same substrate as it is normally used to can make a difference. With dogs this might mean walking on grass areas. Cats can be particularly sensitive to different cat litters. Although it is unfeasible to have the whole range of cat litters in the practice, substrates which closely resemble soft earth (Fuller's earth) may be most acceptable. Many cats find the hard wooden pellet variety uncomfortable. In many practices the kennels used to hospitalise cats are too small. This results in the food and water being placed next to the litter tray. Enclosed litter trays may be more useful; however these must be cleaned frequently.

Recording not only whether the animal has defecated but also some information about the consistency and amount is essential. If the animal is passing diarrhoea, the animal should be checked frequently to ensure the fur around the rear end is kept clean.

Preparation and administration of an enema

Hospitalised animals may become constipated and will need an enema to help evacuate the bowel. The simplest method is to introduce a liquid

solution into the bowel to help loosen hard faeces. Oral solutions may also be given which exert an osmotic affect on the bowel causing water to be drawn into the intestine allowing easier passage of the faeces.

Enemas are usually given to empty the bowel often because of constipation but also prior to bowel surgery or radiography of the lower abdomen. The most appropriate enema solution is warmed water. Soapy solutions used to be commonly administered, however this type of strong alkaline solution can irritate the bowel lining. Liquid paraffin added to the water may also help to ease impacted faeces.

Equipment includes:

- warmed water;
- gloves;
- plastic apron;
- lubricant;
- syringe;
- soft rubber tube or Higginson's syringe;
- funnel or large catheter tip syringe;
- jug for containing and pouring enema solution.

Technique

(1) Gather all equipment together and, if possible, take the animal to an outdoor exercise run/kennel or stand in wash basin or bathtub.
(2) Provide litter tray for cats.
(3) An assistant is helpful to restrain the animal (standing position).
(4) Put on gloves and apron.
(5) Place enema solution into jug.
(6) Lubricate the nozzle of enema tubing and anal ring.
(7) Hold tail up and away and insert tube gently into the anus.
(8) Gently advance the tube, rotating slightly and moving the tube back and forth to advance the tube into the rectum.
(9) Hold the free end of the tube as high as it will allow and attach funnel or syringe.
(10) Pour enema solution into the tube (stand to one side).
(11) Remove the tube after administering required amount of fluid.
(12) Leave animal to evacuate rectum.
(13) Write time of administration and a description of what has been passed on patient's records.

Additional notes

- A gloved, lubricated finger may be gently inserted into the rectum to break up hard faeces.
- Severe impaction may take multiple administrations over several days or a general anaesthetic and manual evacuation.
- Cats may require sedation prior to enema administration; use small amounts with low pressure.

IS THE ANIMAL ABLE TO BREATHE NORMALLY?

It may be very apparent on the physical assessment of the animal whether it has any breathing difficulties. Facial conformation of some breeds can compromise the normal respiration of that animal. Brachycephalic breeds have, by definition, a short nose, an overlong soft palate and a hypoplastic trachea. These can reduce the efficiency of respiration, and obesity, stress and high environmental temperatures can exacerbate the problems. Asking the owner whether the animal routinely snores when asleep can give an indication of potential problems. Age-related problems such as laryngeal paralysis are often aggravated by higher temperatures and increased exercise.

Smell is an important sense for dogs and cats and many cats will refuse to eat if their nasal passages are blocked. Nebulisers can be extremely useful for patients with pneumonia. The nebuliser provides a fine mist of water or saline droplets which help to moisten the respiratory tract secretions. Coupage can then aid the removal of these secretions from the chest.

Respiratory compromise

Caring for animals with respiratory compromise can range from those with a nasal discharge to acute respiratory failure. It is important to remember that finding it difficult to breathe, for whatever reason, is a frightening experience and that these animals must not be stressed further by being placed in a noisy and busy environment.

- Establish and maintain an airway.
- Provide oxygen if necessary (refer to Chapter 40).
- Avoid stress and excessive handling of the patient.
- Closely observe the respiratory pattern, noting changes in depth and character of the breaths.
- If administering medications by mouth, take care not to stress the animal – offer in food if possible.
- Provide assisted feeding if necessary. These patients are often inappetent, but do not overstress by hand feeding.

Tracheotomy tubes

Animals with tracheotomies require intensive monitoring and a high level of care (see also Chapter 40). Tube hygiene is essential and, in the initial stages, the tube may need to be cleaned as often as every 15–30 minutes. The double-lumen tubes have a removable inner tube to enable easier and more thorough cleaning of the tube.

- Clean tracheotomy wound at least daily with antiseptic solution, removing accumulated exudate, and apply povidone–iodine ointment to help protect the skin wound.
- Cover the tube incision site with a keyhole dressing around the tracheotomy tube.

- Observe the animal closely to ensure the tube does not become occluded with bandaging materials, skin folds or bedding. Do not use 'Vetbed' type bedding material, which may clog the tube entrance.
- Ensure the tube is securely attached to the patient and tape in position to prevent dislodgement.
- Instil 1–5 ml of sterile saline down the tube into the trachea every 1–2 hours. This will help to moisturise the trachea, humidify the inspired air and loosen mucous debris.
- Aspirate tracheal secretions by suction using a sterile urethral dog catheter and syringe. Animals with tracheotomies are not able to cough to remove any tracheal secretions.
- Clean the tube frequently. Ideally use a cannulated tube so that the inner tube can be removed for cleaning.
- Prevent self-trauma but use Elizabethan collars with extreme care as they may interfere with the tracheotomy site. Bandage feet if necessary.

IS THE ANIMAL ABLE TO MAINTAIN NORMAL BODY TEMPERATURE?

Maintaining the body temperature within the normal range for that species is an important part of homeostasis. Extremes of body temperature can affect biochemical reaction rates, especially enzymatic reactions. Monitoring the patient's body temperature is part of the VN's role in providing care for each patient. The method is covered in Chapter 3.

There are several factors which can affect the body temperature.

- Time of day: this is usually related to metabolic activity – during the night the metabolism slows and the core body temperature will fall.
- Age: older animals tend to get cold easily as their metabolism slows with age. Very young (neonatal) animals are unable to control their body temperature.
- Hormonal activity: can raise the body temperature slightly.
- Environment: how well animals cope with extremes in environmental temperatures can depend on the thickness of the animal's coat. Working animals that normally live outside in barns may find the temperature in the practice too high.
- Exercise: recent exercise will elevate the normal range of body temperature and this should be taken into account.
- Stress: this can cause an increase in the metabolism, causing a temperature increase.

The thermoregulatory centre is located in the hypothalamus in the brain. It controls the core body temperature by achieving a balance between heat gain and heat loss around a thermoregulatory set point (similar to a thermostat in central heating systems). Heat is lost from the body via radiation, convection, conduction and evaporation. Dogs cool by panting; this is different from breathing quickly as in hyperventilating. There is minimal perspiration through foot pads in dogs and cats. Vasodilation of blood vessels brings blood closer to the skin surface allowing heat loss. Heat is conserved or

gained by constricting the blood vessels, increasing muscular activity through shivering and from metabolic activity (especially from eating).

Hyperthermia or pyrexia

In pyrexia, the circulating pyrogens that occur from infection act on the hypothalamus causing the thermoregulatory set point to increase. This causes the body temperature to increase (shivering and increased metabolism). Heat is also conserved by vessel constriction. Cooling methods should not be used unless the temperature is greater than about 40–41°C.

In hyperthermia, the thermoregulatory set point remains the same and, although the core body temperature is high, the hypothalamus is controlling mechanisms to reduce the temperature. When the body's normal mechanisms fail to achieve adequate cooling, external methods of reducing the temperature should be employed.

Hypothermia

Low body heat occurs from environmental conditions, slow metabolic activity and can be a factor of old age. The thermoregulatory centre is also affected during anaesthesia and the usual attempts to increase body temperature do not work due to the depressant affect of the anaesthetic drugs. In addition the removal of fur and scrub solutions, along with the administration of cold anaesthetic gases all decrease the body temperature. There are a number of warming methods which are discussed in the surgical nursing activities section, Chapter 5.

Keeping patients warm in the wards

There are several different methods for keeping animals warm in the ward. Heat lamps are often used, however it is important to note that this may cause local vessel dilatation leading to further heat loss. Heat pads are useful but the patient must be able to move away from the source of heat if necessary. More effective methods involve reflective blankets and hot air blankets, especially in recumbent animals.

Note

Most of the information refers to mammalian animals. It is important to remember that some animals (reptiles) are poikilothermic: their body temperature depends on ambient temperature. They require certain temperatures in order to feed, so appropriate heating and monitoring of the environment are essential.

IS THE ANIMAL ABLE TO GROOM AND CLEAN ITSELF?

The initial nursing assessment will reveal the normal routine for the animal. Shorthaired dogs are not often groomed by their owners compared to

longhaired dogs. If grooming is done by the owner, then information on the type of brush used and how often should be obtained. Many dogs do wash themselves, most often their paws, and owners may comment on how their dog 'likes to be clean'. Conversely, many dogs may have suffered an unpleasant experience with grooming and may become aggressive. Cats usually wash themselves and often spend some time on their grooming activities. It is usually a good sign to see a cat groom itself in the hospital environment, although in some cases overgrooming can occur as a sign of stress.

Cleaning the animal, removing discharges from eyes, nose, mouth, etc., especially in animals which are unable to groom themselves, will improve the animal's well-being. It is also a useful method of improving the nurse–patient relationship and will help to speed the animal's overall recovery. It also gets the animal used to being handled in the veterinary practice, thereby reducing the patient's fear and anxiety. Grooming is required not only to remove excess hair but also helps identify skin lesions or parasites. For most hospitalised patients, grooming and cleaning should be part of the everyday ward procedure. It is better to spend a few minutes grooming regularly than many hours sporadically. There are exceptions to this, especially when the patient is becoming too stressed. Whilst not a professional groomer, the VN should be aware of some of the different coat types in dog and cat breeds and the grooming equipment most suited to their hair type (Table 4.8). The coat is made up of two main types of hair:

- guard hairs – rigid primary hairs;
- lanugo hairs – soft, thinner hairs.

The type of coat an animal has is governed by the combination of these hairs.

Grooming equipment

Grooming equipment must be used with care. The metal teeth can scratch and bruise the skin very badly. Forced combing or pulling at tangled mats pulls out both live and dead hairs and causes discomfort, pain, irritation and secondary bacterial infection of the skin. It also ruins the coat. Grooming equipment can also act as a fomite, transferring diseases and parasites. Ensure equipment is disinfected between each patient.

- A *slicker* or *carder* may be used to loosen the coat and remove the dead hair. They are useful on smaller hair mats and for general grooming purposes. They may cause injury to areas of the body with sparse hair covering, so use with care.
- *Natural bristle brushes* are considered preferable to nylon bristles because they cause less static electricity, which can cause hair breakage. These brushes are useful on very short hair only because they do not get down to the hair near the skin, so it remains matted whilst the top layer looks fine.
- *Hound gloves* are used on short-haired breeds to remove the dead undercoat and give the coat a shine.

Table 4.8 General care for different coat types in dogs.

Type of coat	Example of breeds	Special tools/equipment	Technique
Long coat	Newfoundland, German Shepherd Dog, Old English Sheepdog, Collies, Siberian Husky, Samoyed, Corgi	Rake. Bristle/wire brush. Fine Resco™ comb	Rake dead hair, comb and brush forward over top and sides, backward over flanks. Fine comb on chin, tail and ears
Silky coat	Spaniels, Afghan Hound, Maltese, Yorkshire Terrier, Setters, Lhasa Apso, Pekingese	Wire brush. Medium and fine steel combs. Bristle brushes	Frequent bathing to prevent mats. Spaniels need stripping every 3 months
Non-shedding curly/woolly coat	Poodles, Bedlington Terrier, Kerry Blue Terrier	Oster™ clippers no. 7, 10 and 15 blades. Natural bristle brush. Steel combs	Clip every 4–8 weeks. Comb and brush to prevent mats
Smooth coat	Dobermann, Retrievers, Boxer, Dachshund, Dalmatian, Beagle	Hound glove. Rubber brush	Rub coat for sleekness and to remove dead hairs
Wiry coat	Fox Terrier, Welsh Terrier, Airedale Terrier, Lakeland Terrier, Schnauzer, Sealyham Terrier	Oster™ clippers no. 7, 10 and 15. Duplex stripping knife, slicker brush, hound glove	Hand strip and brush with slicker and hound glove to remove dead hair
Corded coat	Komondor, Puli	Shampoo – diluted 10:1 in a water spray. Hair dryer	Never clip or comb. Bath if dirty. Squeeze dry – do not brush or rub coat when wet

- *Stripping combs* have a razor blade or serrated metal blade encased in teeth. These are used to help pull out dead hair. The hair is grasped between the thumb and the comb and removed with a twisting motion. Hand stripping or plucking is a technique used by some owners and groomers to remove hairs.

Bathing technique

Most cats do not require bathing and on the whole, only show cats get bathed regularly. Unless the cat likes water, it is usually an extremely stressful time for both the cat and the owner; specific groomer air-drying cabinets would be necessary rather than a hairdryer. Most dogs will have experienced a bath at some point in their life, especially if they have a preference for rolling in smelly substances.

- Before dogs are bathed, their coats should be brushed out and their claws clipped.

- Hair from in between the pads can be trimmed using clippers if possible (safer than scissors).
- Severe mats and tangles should be cut out before they are wet because they become much more difficult to remove after they have become wet.
- The anal sacs should be palpated and expressed if necessary (for technique see p. 73).
- The ears should be examined and excess cerumen removed.
- Pledgets of cotton wool may be placed into each ear before bathing to prevent soap and water entering and causing irritation.
- There is a number of shampoos available from general cleaning shampoos to medicated varieties which can treat skin conditions.

Method

(1) Thoroughly wet skin surface with warm water.
(2) Use a sponge to wet skin around face and difficult-to-reach areas.
(3) Apply shampoo at several points over body.
(4) Gently massage into coat and skin all over the dog until sufficient shampoo is used to produce a good lather (use manufacturer's recommendations).
(5) Take care around the face area to avoid eyes, nose, mouth and ears.
(6) Leave shampoo on for contact time if necessary.
(7) Thoroughly rinse all of the shampoo from the skin using large amounts of warm water.
(8) Squeeze the coat to remove excess water and allow the dog to shake itself.
(9) Thoroughly dry with hand towels.
(10) Gently comb the hair, taking care not to pull mats.
(11) Dry dog using a blow dryer. Take care never to hold the dryer in one place for too long and do not hold too close to the skin.

Nail clipping

Overlong nails can affect the normal walking gait of an animal. Normally these are kept short by the dog walking on hard surfaces. Cats keep their claws sheathed when walking to keep them sharp. Cutting the nails too short can be a painful experience and dogs remember by association.

Technique

(1) An assistant restrains the patient.
(2) Take hold of the foot using the same hand; push each toe to expose the nail fully.
(3) If the quick is visible, place the clippers distal to the quick and cut the nail at an angle with a rapid action.
(4) If the quick is not visible (black claws), apply slight pressure with the clippers at an estimated position.
(5) If the animal reacts badly, reposition the clippers further distally and try again, clipping as before.
(6) Clip each nail in turn, not forgetting to check for dewclaws, these are often very overgrown and can easily be hidden in long-haired animals.

(7) If bleeding occurs, press a piece of cotton wool against the end of the nail and, if bleeding persists, apply a silver nitrate pencil or styptic pencil.
(8) Always ensure that the patient is restrained properly and talked to throughout the procedure to reassure them.

It is advisable to offer treats during and after the process to make future attempts easier.

Emptying anal glands

(1) An assistant is required to restrain the dog, as it may be uncomfortable.
(2) Put on examination gloves.
(3) Apply generous amount of lubricant (liquid paraffin, KY™ gel) to index finger.
(4) Insert lubricated finger into rectum and locate full sacs. They are located at 4 o'clock and 8 o'clock and vary in size (usually about pea size when full).
(5) Gently squeeze contents of the sac dorsomedially between the finger and thumb.
(6) Use cotton wool to wipe sac contents away from around the anus.
(7) Holding the cotton wool in the same hand as the one used to express, remove the glove by inverting it and tying it in a knot to reduce the smell left in the room.
(8) Wipe some animal deodorising spray or wipes around the area to leave a fresher smell.

Clipping beaks

Beak overgrowth is common in small psittacines such as cockatiels and budgerigars. It is usually due to malocclusion, which prevents the beaks wearing down in the normal way. These birds will require their beaks to be trimmed regularly. This may be done using bone forceps, file or fine nail clippers.

Care must be taken not to cut the sensitive structures under the keratin, as this may cause infection or growth abnormality.

After trimming, the beak should be smoothed with emery paper or a nail file.

IS THE ANIMAL ABLE TO MOBILISE ADEQUATELY?

The normal healthy animal is able to move around freely at will. There are various factors which can affect normal mobility including unconsciousness, spinal and orthopaedic problems and debilitating illness resulting in recumbency. The animal needs to have functioning sensory and motor control of the body in order to move around. With the loss of mobility, a number of problems can develop:

- muscle wastage;
- poor tissue perfusion;

- decubital ulcers;
- lung congestion (in recumbent patients).

The veterinary nurse's role

The initial assessment will identify gait abnormalities. Information from the owner may reveal that the animal suffers from arthritis and has difficulty going up stairs or jumping on to high surfaces. It is important to know not only the amount of exercise but the type of exercise, whether the dog has frequent sedate strolls or runs around chasing balls or Frisbees for hours at a time. The nursing care should aim at restoring the animal back to as near normal for that patient as possible.

It is essential that the VN monitors their patients for signs of muscle weakness or reduction in mobility. Veterinary physiotherapists play an important role in assessing patients and creating individual therapy plans. VNs, especially in referral neurology and orthopaedic centres, have specialised in carrying out some of the rehabilitation techniques prescribed by the veterinary physiotherapist. (Currently the title of animal or veterinary physiotherapist is not protected and anyone may practise under this title: it is essential that anyone practising under this title is appropriately qualified.)

Nursing the recumbent patient

Causes of recumbency range from unconsciousness and debilitation to severe spinal trauma. Recumbent patients are often hospitalised for long periods and easily become bored or stressed. They require the intensive nursing techniques described below as well as additional time spent keeping them mentally stimulated. It may be beneficial to call owners in to sit with the animal for short periods each day to help with this aspect of the rehabilitation of these animals.

- Provide a kennel large enough for the patient to lie in lateral recumbency but not so large that the animal is able to drag itself around and cause distress.
- If possible, place the patient in an area of activity to help provide stimulation and relieve boredom.
- Ensure soft, warm, comfortable bedding with lots of padding. Use a foam mattress and thick bedding, e.g. Vetbed and an incontinence pad if necessary.
- Recumbent animals become cold easily so provide additional heating if necessary. Heat pads can cause burns as the animal is unable to move away from the source of heat.
- Keep food and water within reach and hand feed if necessary.
- Feed highly palatable, concentrated and easily digestible food (less faeces!) and consider reducing quantity of food because of the lack of exercise.
- Increase the fibre content of food if the patient becomes constipated.
- Take outside if appropriate, using a towel sling.
- Clean and groom the patient daily. Patients may be unable to do this for themselves and it will help reduce boredom.

- Apply talcum powder on areas where skin may rub such as the inguinal region.
- Massage limbs to aid circulation.
- Clean any decubital ulcers with mild antiseptic solution, dry thoroughly and apply white soft paraffin followed by a 'doughnut' dressing.
- Recumbent patients can be prone to hypostatic pneumonia. Turn patient every 3–4 hours.
- Coupage may be necessary to aid removal of secretions by coughing, perform three to four times daily.
- Encourage sternal recumbency using sandbags, foam wedges or rolled up bedding.
- Prevent urine or faecal scalds by thoroughly cleaning the patient and applying white soft paraffin around the perianal area. Clipping the hair around the perianal area will aid cleaning especially in longhaired breeds. Applying a tail bandage will also help.
- Check bladder frequently to ensure it does not become overdistended. Manually express the bladder if instructed to do so by the veterinary surgeon.

Basic rehabilitation techniques

Basic massage and range-of-movement techniques may be carried out by the VN following advice from the veterinary surgeon and/or a qualified veterinary physiotherapist. VNs should recognise when these techniques may be contraindicated and realise their limitations. The following information relates to basic techniques that can be carried out by the veterinary team, however it is important that the team identify cases that may require further assessment. The following techniques are useful when attempting to restore the animal back to reasonable mobility:

- massage;
- range-of-movement exercises;
- hydrotherapy.

Massage therapy

The main objectives of massage are:

- to relax muscles under tension;
- to increase blood supply (and oxygen) to the tissues and allow removal of waste products;
- to help restore normal function;
- to speed up healing of injured tissues;
- to aid pain relief.

In addition, the relaxing effect of massage helps to build the relationship between nurse and patient.

Indications for massage include:

- tight contracted tendons, ligaments and muscles;
- chronic inflammatory conditions;
- peripheral nerve injuries;
- scar tissue;
- acute and chronic oedema.

Massage is contraindicated and should not be performed in the presence of:

- acute inflammation;
- infection and pyrexia;
- tumours and metastatic disease;
- coagulation problems;
- some cardiac problems.

There are three basic massage techniques:

- effleurage;
- petrissage;
- friction;

Lubricants (e.g. olive oil, soft white paraffin or baby powder) may be used to soften the skin and reduce friction, although they can be difficult to remove from the fur. These agents should not be applied to wounds. Whichever type of massage is most appropriate, always start each session with effleurage as a warm up to the main session.

Effleurage

This is a gentle massage technique, which precedes the other two types. Effleurage accustoms the animal to touch and warms up the underlying tissues. The hands work using a light or heavy stroke with uniform pressure. The patient is massaged from distal to proximal on the limbs in the direction of venous return. A light stroke at a rate of 15 strokes per minute is applied for a sedative effect. A heavy stroke at a rate of five strokes per minute is applied to enhance draining of lymph channels. This part of the session should last approximately 10 minutes to prepare the patient for more extensive massage using one or more of the techniques below, depending on the animal's condition. In the first 24–48 hours after spinal surgery this should be limited to 3–5 minutes.

Petrissage (kneading or compression)

This is used primarily on muscles to enhance circulation and stretch muscles, tendons, adhesions and contractions. The muscle is compressed from side to side as the hands move up the muscle, always in the direction of venous return. The pressure should be gentle and never forceful enough to cause pain.

The amount of time spent on massage will vary in each case but the following may be used as guidelines (times stated are for each limb).

- To reduce limb oedema: 5–10 minutes, two to three times daily (until swelling goes down).
- Spinal patient with voluntary movement: 10–15 minutes, two to three times daily.
- Spinal patient with no voluntary movement: 15–30 minutes, two to three times daily.
- Spinal patient with no voluntary movement and stiff joints/muscle spasm: 15–60 minutes, three to four times daily.
- Orthopaedic/spinal patient requiring joint mobilisation: 5–10 minutes prior to joint flexion/extension.

Friction

Used to aid the absorption of local effusions and loosen superficial scar tissue and adhesions. Skin is moved over the underlying tissue in small, circular, rhythmic motions while pressure is applied across the tendon or adhesion to encourage collagen formation. Should only last 3–7 minutes.

Range-of-movement techniques

Passive range-of-movement techniques

These are used to retain and/or regain joint mobility. The limb is held above and below the affected joint and the joint is put through its normal range of motion. This should be done no more than ten times on each joint three to five times a day. Never perform passive movement on a joint that has not been warmed up first and always prepare the area using effleurage first to warm up the joint and prevent pain on manipulation. Each time the joint is moved, it should be held in flexion/extension for 5–10 seconds. Very stiff joints may be held in position for longer. This should be repeated three to five times each joint per session. Up to five sessions may be performed daily if necessary.

Active movement

Active movement involves voluntary activity by the patient. It can be started with gentle pulling on the toes and encouraging the patient to pull them away. For paraplegics, a towel or harness is looped under the abdomen and supported, whilst encouraging the patient to weight bear on the hind limbs. The patient may be walked around to maintain muscle tone in the front limbs. The total time spent on harness walking will depend on the individual case, but generally 10 minutes three times daily is adequate. Encouraging the patient to sit and stand increases joint mobility and hind limb strength. Stair climbing can also be useful to increase muscle strength. The exercises prescribed by the physiotherapist are designed to improve the following:

- strength;
- endurance;
- flexibility;
- balance and proprioception.

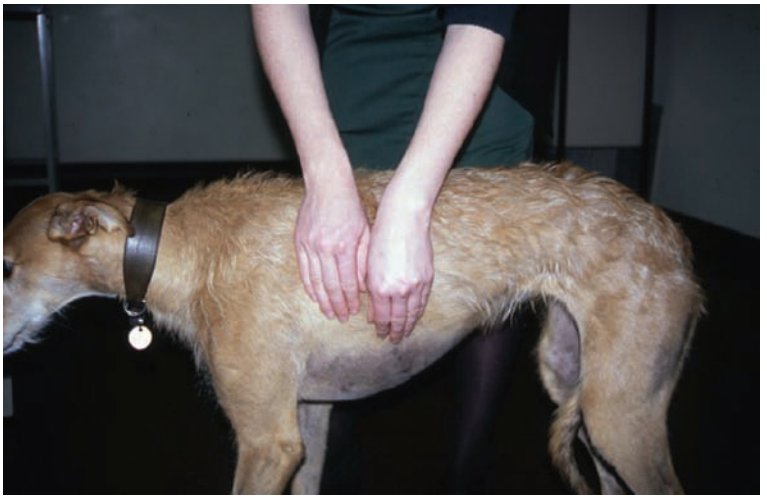

Fig. 4.7 Coupage.

The nurse must monitor and record the animal's progress. The initial goal with recumbent patients is to get them to weight bear on their limbs. With the animal supported in the standing position the muscles are being used and strengthened. Paraplegics can require a lot of nursing care and it may seem that progress is slow. However, with appropriate care planning, goals can be seen to be achieved.

Coupage

This is used to clear any secretions from the airway (hypostatic congestion) by gentle percussion of the thorax using cupped hands (Fig. 4.7). Always work from the caudal end of the chest to the cranial end, this will encourage coughing and the removal of secretions. Always percuss both sides of the thorax. Coupage should be performed *gently* and not cause lung damage.

Hydrotherapy

The physical properties of water buoyancy and hydrostatic pressure provide support. The patient should be immersed into a bath or whirlpool that is just too deep for the animal to stand and be supported by the handler while swimming (Fig. 4.8). Water movement, by means of a shower head attachment, can be added to create a massage effect. The water temperature should be 38.8–40.5°C (102–105°F). If using small baths, povidone–iodine or similar can be added to the water to help prevent any skin infections from decubital ulcers or urinary scalds. In larger pools dogs may be encouraged to swim around with the use of water toys. Buoyancy jackets help the less able swimmer feel more secure in the water. Animals must never be left alone in

Fig. 4.8 Hydrotherapy.

the water. Some animals will need reassurance and encouragement to ease fear. Always dry the patient thoroughly following bathing.

Cold therapy

Application of cold to an area causes vasoconstriction, which helps to reduce inflammation and provide mild analgesia. Cold tends to penetrate deeper and last longer than heat.

Cold therapy is indicated to:

- reduce/prevent oedema;
- decrease postoperative swelling;
- reduce muscle spasms.

It is contraindicated in:

- open wounds;
- circulatory problems.

This therapy is usually used as a first aid measure immediately following a traumatic injury. Various cold packs may be used including:

- commercial cold compression units;
- packets of frozen vegetables;
- a latex glove filled with very cold water;
- pieces of ice placed into leak-proof plastic bags.

All types of ice packs should be wrapped inside a towel to protect the animal's skin and should be applied for 10–15 minutes.

Heat therapy

Heat therapy is useful for its analgesic properties. Heat helps to reduce muscle stiffness and muscle spasms and promotes wound healing. Its application is also useful:

- postoperatively;
- for relieving stiff joints;
- for relieving muscle stiffness;
- following trauma.

It should not be used for patients with decreased sensation, e.g. neurological patients or in acute inflammatory conditions.

Methods of applying heat include:

- commercial heat packs;
- hot water bottle wrapped in a towel;
- heat pads;
- drip bags heated in the microwave;
- 'wheat bags' heated in the microwave;
- latex gloves filled with warm water;
- infra-red lamps.

The length of time the heat should be applied depends on the condition and the methods used.

Contrast bathing

This technique is usually used after time has elapsed so that cold therapy will no longer be effective. It is an excellent method of increasing circulatory flow, decreasing swelling and contusions, and speeding the elimination of tissue exudate. An ice pack is applied for 3 minutes followed by a heat pack for 3 minutes. This procedure is repeated for 20–30 minutes.

IS THE ANIMAL ABLE TO SLEEP AND REST ADEQUATELY?

Sleep is essential to all animals. Some species, such as small rodents, sleep at different times of the day and allowances should be made for this when deciding where they are to be hospitalised. Most dogs and cats sleep on average about 10–12 hours a day. Many owners are probably not aware of the total time their pet is asleep during a 24 hr period. The stressful environment of the hospital can prevent animals getting enough sleep.

There are two processes involved in controlling sleep. One is the response to how long the animal has been awake and ensures sleep occurs so that restoration of the body can happen. The second process involves the timing of sleep during a 24-hour period – the circadian rhythm. Secretion of various neurotransmitters at certain times of the day ensures sleep occurs usually at the same time each day.

Sleep is necessary to recovery and all hospitalised animals must have the appropriate environment to enable this. Ward lights must not be left on all

night. Having a room between each of the wards allows procedures to be carried out on patients without disturbing the others.

IS THE ANIMAL ABLE TO EXPRESS NORMAL BEHAVIOUR?

In order to assess this aspect the VN must have a good understanding of the normal behaviour of the particular species they are caring for. Consideration of the five freedoms stated previously helps as a framework from which to start.

Cats

Cats tend to be solitary predatory animals, although recent research has shown they do form 'buddies', often with related individuals. Being solitary predators it is important to them not to get hurt. When threatened they tend to use 'freeze' or 'flight' behaviours rather than 'fight'. However if cornered, this may be their only option. Keeping many cats in a household can be extremely stressful to an individual cat and various behaviours appear as a result. Territory tends to be important to cats. They have various marking behaviours to outline their territory. These include:

- rubbing;
- scratching;
- spraying;
- urinating and defecating.

Many of the 'unwanted' behaviours occur because the cat is trying to reinforce their scent in order to feel less stressed.

Pheromones play an important part in helping the cat feel less stressed and these should be employed appropriately in the hospital environment. Having a separate cat ward is important, and it should be ensured that the kennels do not face each other.

Dogs

Dogs are pack animals and their social structure means they usually enjoy company of other dogs. Posture and facial expression is an important part of communication between dogs. It is essential that the VN can also read the signals that may be shown by an individual dog. There is some debate over the origin of the dog and whether their behaviour is identical to that of wolves. There are many similarities but also some differences. It is important to remember that domestication and evolution have played a part in the development of the dog.

Rabbits and other small pets

Rabbits are increasingly becoming a popular pet and, with domestication, behavioural problems can occur. Rabbits are prey animals and become

stressed if not provided with areas to hide away or are constantly exposed to predator smells. In nature, rabbits spend long periods of the day eating and foraging for food. Concentrated pellet food is convenient but does not provide the animal with the stimulation of digging and working for their food.

Small rodents also have the need for an enriched environment providing them with stimulation to prevent behavioural problems. Reptiles should have appropriate hospitalisation with gradients of temperature.

Sexual behaviour

Knowledge of whether the animal is entire or neutered will be on the patient's records. This information may not be known about newly acquired animals from rescue centres. The VN should be able to recognise signs of females being in season and accommodate them appropriately in the practice – not near entire males of the same species. Changes in the animal's behaviour may be related to sex hormones.

Environmental enrichment for hospitalised patients

Spending long periods in the hospital can be frustrating for normally very active animals. Mental stimulation is often important to help speed recovery, improve mental attitude and help prevent boredom. Toys from home can help the patient settle in more easily – it is essential these do not get lost in the practice and a record of the type of toy and description must be made at admission. Using feeding toys where the animal has to spend time working for their food can help. Freezing a portion of their food in a Kong™ toy can keep a normally active dog busy for quite some time. For recumbent animals the highlight of their day is often meal times, but the food does not last long. Simple toys from strips of paper towels and toilet rolls can amuse kittens and young cats. Small animals such as rabbits and wildlife may be terrified in the hospital environment and may need boxes and hay to hide in.

Training commands

During the initial nursing assessment it is useful to ascertain whether the dog has any specific training commands, especially for urination and defecation purposes. For working guard dogs there may be certain commands which must not be used!

HANDLING AND RESTRAINT OF SMALL ANIMALS

Dogs, cats and other small animals can inflict serious injuries if not handled correctly. It is important to learn handling techniques but also to be aware of signals of aggression and be able to deal with the animal appropriately. Understanding the body language of animals is essential for anyone handling them on a regular basis. *Always* observe the animal before approaching and expect the unexpected!

Indications for restraint include:

- to allow diagnostic procedures;
- to allow therapeutic procedures (administration of medicines);
- to prevent injury to itself and to others.

Approaching the animal

(1) Observe behaviour and assess the animal, and ask the owner.
(2) Approach quietly and confidently – using its name and talking in a reassuring manner.
(3) Bend down to the animal's level.
(4) Offer closed fist for the animal to sniff if no signs of aggression are seen.

Remember the following points.

- Animals often behave better away from the owners.
- When taking a dog from an owner, take the owner and dog into a room away from the waiting room.
- Use a slip lead as dogs may often slip their collars.
- Ask the owner to leave the room rather than dragging the dog from the room.

Aggression

Causes of aggression in animals

It is important to realise that animals which are normally placid react differently when in pain, in a strange environment or scared. Assess the situation carefully. Causes of aggression include:

- possessiveness – territorial, food, owner;
- fear;
- pain;
- maternal protection;
- breed predisposition (such as guard dogs and working dogs);
- too much restraint (especially in brachycephalic dogs);
- dominance.

When handling an aggressive cat or dog, remember:

- be firm;
- be confident in handling.

Equipment for controlling aggressive dogs

It is essential that a nurse can apply this equipment correctly – if there is any doubt of the dog's temperament, muzzles should be used to prevent injury to personnel. Dogs can be trained from an early age to accept a muzzle. Suitable equipment includes:

- muzzle, e.g. Mikki™, Baskerville™, bandage tape muzzle;
- dog catcher.

Equipment for controlling aggressive cats

There are various pieces of equipment that can be used to control an aggressive cat. Make sure you know how to use them correctly. Suitable equipment includes:

- thick towel to retrieve the cat from the kennel and to wrap the cat in;
- Mikki™ muzzle;
- crush cage;
- leather gauntlets;
- cat bag;
- cat catcher (as a last resort! The cat *must* be supported correctly).

Moving animals around the practice

Cats

Cats should always be transported around the practice securely in a basket. A cat that is carried in the arms may easily be frightened and escape!

Dogs

When taking a dog from one area to another ensure it has a secure lead attached. Slip leads are routinely used as the dog cannot easily 'back' out of it!

Small mammals

Small mammals are often transported in cat carriers. Make sure they cannot escape through the wire mesh or air holes. Rodents are best not transported in cardboard boxes, as they will often eat their way out. See Table 4.9 for details about handling small animals.

Birds

Birds are often brought to the surgery in their own cages. They should be placed in a quiet, darkened area to keep them calm and reduce stress. Birds that arrive in small cardboard boxes are fine for a short period only – they should ideally be transferred to a larger cage.

Carrying and lifting

Many VNs suffer from back problems from lifting incorrectly. Attempting to lift a dog that is too heavy may result in injury to the VN and the dog. Animals often struggle if they do not feel secure when they are being carried. Always ensure that there is adequate control over an animal so that no one gets injured. When lifting large dogs (greater than 25 kg), two or more people are required. Always ensure the dog's head is controlled and that the animal is well supported. For smaller dogs it is sufficient to ensure the dog's head is controlled and the body is well supported.

Table 4.9 Methods of handling and restraint for small mammals, reptiles and birds.

Animal	Method of restraint
Rabbits	Handle carefully and quietly. If handled roughly, rabbits may easily damage their spine and become paralysed by kicking out with their hind legs. DO NOT pick up by their ears
Guinea pigs	Place one hand around the shoulders and support the hind quarters. Most guinea pigs are reasonably tame – but do not frighten or startle
Rats	Rats rarely bite unless frightened or in pain. Pick up by placing a hand around the shoulders. Position the thumb under the mandible to prevent biting. 'Scruffing' rats causes considerable distress and do not pick up by the tail
Mice	Mice may bite if frightened – handle with care. Scruff gently and grip base of tail with 3rd or 4th finger
Gerbils	Pick up by gently cupping hands around the animal. Restrain by holding base of tail. If more restraint is required gently hold by the scruff. Take care that it doesn't jump off the table
Hamsters	Handle with care! Hamsters can inflict painful bites. Pick up by gently cupping hands around the animal. Further restraint may be by scruffing gently
Chinchilla	Hold firmly around the shoulders and support the body with the other hand. Chinchillas escape easily leaving you with handfuls of fur!
Ferrets	If handled frequently ferrets are quite tame. Grasp around the neck/shoulders with one hand whilst supporting the body and hind legs with the other. When they bite they usually hang on tight!
Budgies	Hold head gently between 1st and 2nd finger. Take care not to asphyxiate bird! Offer a pen or something they can peck on rather than your finger.
Large birds	Large birds can be handled more easily by wrapping them in a large towel or by wearing thick gloves. Ensure their wings are secure to avoid them flapping around
Snakes	More exotic species such as snakes and lizards are often seen in practices which specialise in this area of animal care. There are strict licensing laws covering the possession of many exotics and so it is likely that the owner is familiar with the handling of their own pet. However it is important to be aware of the different methods. Non-venomous snakes may be handled by grasping the head with the thumb and middle finger while the index finger is placed on top of the head. Always support the rest of the snake's body. Venomous snakes should only be handled by experienced personnel using snake hooks, plastic tubing or a pillow case
Lizards	Place one hand around the shoulders and the other across the reptile's back around the hind limbs. Restrain the head of aggressive species. Never lift or catch by their tail as many shed their tail as a mechanism of escape
Tortoises and other chelonian	Hold firmly by the middle of the shell (box turtles can close their shells at the front and back and trap a finger). Beware of snapping turtles. Remember many species carry *Salmonella* and other zoonotic diseases. Always wash your hands before and after handling

Restraint

Dogs

It is essential to be able to restrain dogs in different positions to be able to undertake various diagnostic tests or examinations. Dogs may be restrained in lateral recumbency or a sitting position. The VN must always ensure they have adequate control over an animal so that no one gets injured, and must ask for help if needed (i.e. 'backstop').

Cats

The usual method is to 'scruff' the cat, however this can be very stressful and will upset a normally placid animal, so the VN should always assess the level of restraint required. In a lot of cases just cupping the cat's head in the VN's hand whilst restraining the rest of the body with the other arm is enough. Remember to hold on securely to the front legs. With more aggressive cats, 'wrapping' the cat in a thick towel or using a 'cat bag' may be necessary.

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Chapter 5

Nursing Notes for Surgical and Medical Conditions

Animals with different conditions and diseases have many different nursing care requirements and it is important that the VN is aware of each patient's specific needs and how to go about caring for them during hospitalisation. Thorough assessment, close observational skills, being aware of potential problems and being able to provide a high level of nursing care will help to ensure patient comfort and to speed up rehabilitation, thus reducing lengthy periods of hospitalisation.

As described in Chapters 3 and 4, a thorough daily assessment and measurement of vital signs is essential. A temperature, pulse and respiratory rate measurement should be obtained at least once a day for every hospitalised patient. In addition to this, the following care plans are designed to give a quick point of reference for the additional care required for some of the common surgical and medical conditions. It should be remembered, however, that each patient's needs are unique and the VN must discuss appropriate care plans for every hospitalised animal with the veterinary surgeon in charge.

CARE PLANS FOR GENERAL CONDITIONS

Geriatrics

Geriatric patients admitted to the practice require particular attention in order to try to maintain their normal routine. The VN should obtain information from the owner regarding the patient's normal lifestyle and treatment at home. Older animals frequently suffer from multiple diseases and the VN should implement appropriate nursing care for each condition. In addition, any existing medication plans must be continued, checking first that they are not contraindicated with any new treatment being prescribed.

- Ensure that water is available ad lib and monitor and record intake quantities.
- Feed a high-quality protein to help reduce muscle mass loss, but not high levels of protein in cases where there is existing kidney disease.
- Give small, frequent, highly palatable meals rather than overloading the digestive system and organs with one or two large feeds per day.
- If necessary, reduce the quantity being given to help reduce obesity.

- Provide soft, warm, comfortable, easily washable bedding for comfort and to help prevent decubital ulcers over bony prominences.
- Additional heating may be required due to poor circulation.
- Keep away from draughts.
- Ensure frequent but short trips outside to help reduce incontinence problems and reduce stiffness.
- Groom the patient daily as geriatric patients are less likely to clean themselves.
- Handle gently and take care as older animals are more likely to become irritable and snap at handlers.

Neonates

If the dam dies, loses her milk supply or becomes hypocalcaemic, the neonates will have to be hand reared. If the young have not already received some colostrum, try to milk some from the mammary glands of the dam (provided it is not contaminated with drugs or toxins) and give to the young in one of their first feeds. Orphaned offspring are at a higher risk of getting infections because they have often not received any colostrum and therefore do not have any maternal antibodies. Thorough hygiene is essential.

- Always wash your hands before handling.
- Place orphans into an incubator if available or use infrared lamp, hot water bottles or heated corn pad. Take care not to cause burns. Remember not to let the temperature drop at night time. The ambient temperature should be 24–27°C (75–80°F) for the first 10 days, reducing gradually to room temperature by week four.
- Always sterilise feeding equipment before each use.
- Use commercial milk substitute. Cow's milk and goat's milk are not suitable. Examples of milk substitutes include Lactol, Pedigree Instant Milk Substitute, Welpi, Cimicat and Whiskas Instant Milk Substitute.
- Use Catac or Hagan pet feeding bottle in preference to stomach tube, which does not satisfy their natural sucking reflex.
- Calculate required amount according to manufacturer's instructions.
- Sterilise feeding equipment in Milton sterilising fluid.
- Warm food to body temperature before feeding (39°C).
- Feed every 2–4 hours throughout the day and night for the first few days then reduce to every 4 hours.
- Hold the neonate in sternal recumbency during feeding to encourage the natural paddling movement of limbs.
- After feeding, stimulate the young to pass urine and faeces by rubbing their abdomen and perianal region with a piece of warm, damp cotton wool until they void both (record all motions – bottle-fed young are prone to constipation).
- Clean the animal using warm, damp cotton wool – wipe around their mouth and nose to remove dried milk and clean around the rest of the body.
- Some humidity is important and can be provided by placing a piece of damp towelling near their box.

CARE PLANS FOR PATIENTS WITH MEDICAL CONDITIONS

Patients admitted to the hospital for a medical work-up will frequently undergo many diagnostic tests before a therapeutic plan is worked out specifically for them. The VN may be responsible for obtaining and processing laboratory samples (see Section 7) and taking radiographs (see Section 6) as well as using other diagnostic tools.

Once a diagnosis has been established, the nursing care should be tailored to meet the individual requirements of the patient. The following care plans highlight nursing considerations for some of the common medical conditions seen in hospitalised patients. As previously mentioned, it is especially imperative that vital signs are monitored frequently and the patient clinically assessed so that changes and deterioration can be picked up as quickly as possible.

Chemotherapy

Cancer patients require a high level of nursing attention and skill. These animals are often hospitalised for long periods and easily become depressed and anorexic. Chemotherapy patients have impaired immune function and are therefore more susceptible to infection, so good hygiene is essential.

The drugs used for chemotherapy are highly cytotoxic. Most chemotherapy agents are cytotoxic, mutogenic and teratogenic and present a real danger to operators. The VN must exercise extreme care at all times when they are used and during the nursing of recipient patients.

- Wear two pairs of gloves at all times when handling cytotoxic drugs and during any time when dealing with the animal.
- Avoid cutting or breaking tablets if possible and wear a mask if this must be done.
- Use pill cutter when cutting cytotoxic drugs. Label cutter and only ever use with cytotoxic drugs.
- Wear disposable plastic aprons.
- Reconstitute cytotoxic drugs in a plastic tray (e.g. cat litter tray) to contain any spillages and ease cleaning.
- Reconstitute cytotoxic drugs in a well ventilated room but ensure doors and windows are closed to prevent draughts.
- Cover all cuts and scratches.
- Wear protective goggles.
- Wear a good-quality surgical face mask if reconstituting cytotoxic powders.
- Use insulin syringes or luer-lock syringes and giving sets to prevent accidental leakage of cytotoxic drugs.
- Wrap a sterile gauze pad around the top of any glass vials when opening them to prevent spillage.
- Cap all needles before expelling air from the syringe or expel into the vial or ampoule.
- Wrap sterile gauze pad around the needle when removing it from an injection port to help prevent needle-stick injury.
- Clearly label all drip bags containing cytotoxic agents.

Diabetes

Hospitalised patients with diabetes are likely to be either unstabilised and admitted for monitoring, or stabilised patients coming in for surgery. The VN must be able to recognise signs of hypoglycaemia (shivering, weakness, tachycardia and seizures) and the signs of hyperglycaemia (diabetic coma).

- Closely observe clinical signs and maintain accurate and thorough records regarding appetite.
- Monitor and maintain accurate and thorough records of blood glucose testing and administration of insulin.
- Adhere to a strict routine for feeding time and quantity, time of insulin administration and exercise.
- Encourage the patient to eat. In the event of inappetance an animal is more unlikely to eat a prescription diet, but it is important to get them eating something so try any palatable food.
- Diabetic animals undergoing anaesthesia are starved as usual but given half their usual dose of insulin on the morning of the procedure. Blood glucose should then be tested.
- If blood glucose levels are low then a glucose saline infusion may be administered during anaesthesia and blood glucose levels retested following surgery.
- During stabilisation, blood glucose should be monitored every 4 hours.
- When administering insulin, gently agitate the insulin bottle before withdrawing the insulin and do not wipe the rubber stopper with isopropyl alcohol.

Diarrhoea

There are many causes of diarrhoea. Diarrhoea may be secondary to some other organ dysfunction such as renal failure or hepatic disease, in which case further nursing skills must be initiated. Other causes of diarrhoea are infectious and/or zoonotic, and until a diagnosis is established it is advisable to barrier nurse the patient. Complications include dehydration and weight loss.

- Withhold food for 24 hours.
- Offer small amounts of oral electrolyte solution or water and monitor fluid intake.
- Assess hydration status frequently for early detection of dehydration.
- Manage fluid therapy, check patency of lines and rate of administration.
- Record frequency, consistency and colour of diarrhoea.
- Ensure the patient and kennel are kept clean.
- The VN must take particular care with their own personal hygiene precautions.

Heart disease

Animals with acute cardiac problems require close observation and as little handling and stress as is possible in the hospital environment. The VN should

also remember that some older patients will have cardiac disease even though they have been admitted for another reason.

- Do not stress the animal, handle as little as possible and keep it in a quiet area, preferably away from other animals. A towel partially covering the kennel door may be of benefit.
- Monitor the patient as frequently as advised by the veterinary surgeon. It will help to detect early any deterioration in the condition.
- Restrict exercise and cage rest. Provide short trips outside to urinate, especially if diuretics are being administered.
- Keep the patient warm as peripheral circulation may be compromised.
- Prepare and provide oxygen therapy if the patient becomes cyanotic or dyspnoeic.
- Ensure that the correct drug doses and administration times are applied.
- Provide an appropriate diet, such as low sodium, and restrict calories if the patient is obese.
- Be confident in setting up and using cardiac monitoring equipment such as electrocardiograms, pulse oximeters and blood pressure monitors.

Liver disease

Hepatitis or inflammation of the liver is used to describe many diseases of the liver such as drug toxicity, viruses, infectious disease and neoplasia. Clinical signs (except jaundice and ascites) are wide ranging and vague and may not be present until 80% or more of hepatic tissue is damaged.

Removal of the causal agent may not be possible, so the treatment and care plan may be supportive and symptomatic.

- Provide a stress-free environment and cage rest for the patient to minimise discomfort.
- Handle the patient carefully and gently: patients with liver disease often have severe abdominal pain.
- Assist feed the patient if necessary. Many patients with hepatic disease become inappetent and anorexic.
- Provide a diet high in calories, but try to avoid a high-protein diet, as metabolism may be impaired. Fat content should be minimised.
- Supplement vitamins (such as vitamins B, C and K) as prescribed by the veterinary surgeon.
- If diuretics are being administered, provide adequate opportunities to urinate.
- Observe for clotting disorders, seizure activity, jaundice and ascites.

Pancreatitis

Causes of pancreatitis include, amongst others, infection, duct obstruction, trauma, drugs and hypercalcaemia. Acute pancreatitis is a life-threatening condition and permanent impairment of endocrine and/or exocrine functions can occur as a result.

- Clinical signs include vomiting, anorexia, diarrhoea and depression. Refer to the appropriate care plan in this chapter for details of specific nursing protocols. The aim of the care plan is to alleviate the clinical signs and provide supportive therapy whilst enabling the pancreas to return (as much as possible) to its normal function.
- Withhold food and water for 3–4 days to avoid stimulation of the pancreatic enzymes and allow the pancreas to rest.
- Maintain intravenous fluid therapy treatment at rates to replace fluid deficits and maintain hydration during the starvation period.
- Administer analgesia as prescribed by the veterinary surgeon to relieve abdominal pain. These patients will often demonstrate abdominal pain by adopting the 'prayer' position.
- When there has been no episode of vomiting for 1 or 2 days, small amounts of water can be introduced.
- If water is tolerated, small amounts of food may be given. A high-carbohydrate (rice, pasta, potatoes), low-protein and low-fat diet should be given to help prevent a relapse. On the first day give only one third of the usual calorific requirements, increasing to two thirds on day two and full amount by day three.
- Monitor blood and urine glucose for the presence of diabetes mellitus.
- Monitor patient for development of disseminated intravascular coagulation (DIC), e.g. platelet counts and prothrombin time.

Renal disease

Signs of renal disease occur when there is loss of function of more than 75% of the nephrons and renal failure has occurred. The kidneys are no longer able to maintain the regulatory, excretory and endocrine functions and, as a result, fluid, electrolyte and acid–base imbalances occur. Treatment plans for chronic and acute renal failure vary slightly, but both include elimination of the underlying cause (if possible) and supporting the patient until renal function has been restored.

- Provide a quiet, stress-free and warm environment for the patient.
- Administer fluid therapy at prescribed rates to replace fluid losses and maintain hydration.
- It is essential to monitor urine output as this provides one of the most important indicators of renal function in the severely compromised patient. In the event of anuria, fluid therapy may be discontinued.
- Observe nursing care plan for those patients with indwelling urinary catheters and urine collection systems.
- Feed little and often and give a diet of reduced protein of high biological value to minimise metabolic wastes and keep blood urea nitrogen levels as near to normal as possible.
- Provide unlimited access to water.
- Assist feed if the patient is anorexic.
- Obtain blood samples for urea nitrogen, creatinine and potassium concentration as requested by the veterinary surgeon.

- Provide opportunities for the patient to urinate frequently; a clean litter tray for cats and frequent trips outside for dogs.

Seizures

Most seizures result in unconsciousness but are for a short duration of time; however, status epilepticus refers to continuous seizure activity and is classed as an emergency situation.

- Hospitalise the patient in a quiet stress-free environment with minimal stimulation.
- If possible, darken the room and partially cover the front of the kennel door with a blanket, but ensure that observation of the patient is still possible.
- Place an intravenous catheter to ensure easy vascular access in the event of a seizure.
- Prepare antiseizure medication (usually diazepam) for rapid administration (do not draw up into the syringe until required).
- Do not try to restrain the patient during a convulsion.
- Monitor time and detail any seizure activity.
- Maintain supportive treatment such as fluids and collect blood and urine samples as required for diagnostic and monitoring purposes.

Vomiting animals

There are many things that cause the patient to vomit and treatment varies depending on the specific cause. However, the aim of therapy apart from removing the initiating cause is to control the vomiting episodes and avoid associated complications such as dehydration, electrolyte and acid–base imbalances and abdominal pain. It is important to remember that clinical signs of dehydration are not evident until the animal is quite significantly dehydrated.

- Observe for signs of nausea and administer anti-emetics as prescribed by the veterinary surgeon.
- Observe the patient for signs of dehydration.
- Observe and record contents of vomit.
- Offer small amounts of water or electrolyte solutions, but do not leave bowl in kennel.
- Handle gently, abdominal area may be very painful.
- Syringe fluids carefully to avoid aspiration pneumonia.
- Clear vomitus away from animal's fur and kennel as soon as it is expelled.
- Withhold food for 12 hours.
- Assess hydration status and initiate intravenous fluid therapy if necessary.
- If and when the vomiting has ceased, small amounts of bland food such as fish, chicken or appropriate prescription diet may be given three to four times a day. Initially, give only a quarter of the daily allowance, and

increase gradually over the following days until normal feeding amounts can be given.

CARE PLANS FOR POSTSURGICAL PATIENTS

Following surgery, the patient is returned to the ward area and the veterinary nurse must provide aftercare aimed at preventing potential postoperative complications resulting from either the surgery or anaesthetic. More information with regard to postanaesthetic care is given in Section 5 with specific regard to management of hypothermia and analgesia. The following care plans are designed to highlight potential problems and specific nursing care relating to common surgical procedures.

Respiratory and circulatory complications and fluid and electrolyte imbalances are potential problems following any surgical procedure and the VN should be able to recognise them and act appropriately.

Postoperative care – general guidelines

Postoperative care and observation of the patient are essential parts of the whole surgical procedure and should never be underestimated. The VN must be confident about which parameters to monitor and be aware of the common complications associated with each type of surgical procedure and each individual patient. Routinely, a temperature, pulse and respiration rate must be obtained and recorded every 15 minutes until a return to normal is seen. The patient should never be left unattended whilst the endotracheal tube is in place. Feeding after surgery is important in small exotics to prevent gut stasis. It is now thought that feeding following gastric/intestinal surgery is more beneficial than starving for a further 24 hours. Diabetics should be fed soon after recovery to maintain glucose levels.

Hypothermia

Hypothermia is extremely common following anaesthesia and surgery due to the combined effects of the presurgical clip and scrub, cold anaesthetic gases administered via a non-rebreathing circuit, cold operating rooms and tables, a reduction in muscular activity during anaesthesia and an anaesthetic-induced decrease in the basal metabolic rate. It is also the most common cause of bradycardia following surgery. Reduction or preferably prevention of hypothermia is an important factor for a rapid recovery from anaesthesia and to prevent patient discomfort. Hypothermia slows down the metabolic rate and therefore the metabolism of anaesthetic drugs.

External heat will be required in the hypothermic patient in the form of:

- blankets;
- bubble wrap;
- heat pads;
- lamps.

It must be remembered that the recumbent animal is unable to move away from any heat source and is at risk of burning. Direct contact with heat pads, hot water bottles and skin must be avoided. During surgery, some simple techniques can be employed which will help to reduce hypothermia. These include:

- warming all intravenous fluids;
- warming bags of saline used during open body cavity flushing;
- keeping surgery time to a minimum;
- maintaining a warm ambient operating room temperature (23°C);
- using an in-circuit water vapour trapping device (Thermovent 600, Portex Ltd) placed between the endotracheal tube and the anaesthetic circuit.

Rapid increases in body temperature and hyperthermia should be avoided. Central nervous system damage can occur as a result of dehydration, hypotension and hypoxia. Frequent monitoring of peripheral and core temperature is therefore essential, and once a return to normothermia is achieved, heat sources should be removed or turned off. As a guide, the patient may be warmed by at least 0.5°C/hour. Measurement of peripheral temperature is useful to provide information about the quality of peripheral perfusion. Under normal conditions in normal room temperature, peripheral temperature (measured between the third and fourth rear digits in the foot) is 2–5°C lower than core temperature. Temperature differences greater than this indicate vasoconstriction and poor peripheral perfusion associated with shock.

Postoperative pain

Observation of the patient's behaviour following surgery is important to detect signs of pain. It is helpful to observe the animal's general behaviour before surgery so that the individual nature of each patient can be considered in postoperative pain assessment. It is well known that it is better to prevent pain than to treat it and that analgesic drugs are significantly more effective if administered before the onset of pain, for example as part of the premedication and during anaesthesia. Animals that are stressed and anxious will be more sensitive to pain. Gentle handling at all times during the patient's stay in the hospital and allowing the animal to recover in a quiet, dimly lit area will help to reduce stress.

At different stages in the recovery period, an animal may show different clinical signs of pain. In the initial stages there may be obvious signs such as howling, restlessness and thrashing around in the kennel. As the patient regains consciousness, clinical signs may become less obvious but may include some behavioural changes, such as anorexia, failing to drink and groom and adopting an unusual posture. Opioid analgesics provide the most effective means of pain control in small animals. More information about the action, effectiveness and appropriateness of these drugs is discussed in Section 5. Non-steroidal anti-inflammatory drugs (NSAIDs) only relieve pain of low to moderate intensity. Care must be taken when using NSAIDs, especially in patients with renal, hepatic or cardiac dysfunction. Aspirin may decrease platelet aggregation and therefore cause increased bleeding times. It should not be used as preoperative analgesia for animals.

Postoperative wounds

All surgical wounds must be examined daily for evidence of haemorrhage, inflammation, infection or patient interference. Surgical wounds do not usually show evidence of infection immediately following surgery. However, the wound is most susceptible to contamination in the initial postoperative period. Usually the patient is hospitalised during this period and so general ward and personal hygiene are essential to help prevent postoperative wound infection.

- Wash hands and wear sterile gloves before examining the wound or changing the dressing.
- Protect the wound from external contamination with a suitable dressing material (see Section 4) for the first 48 hours.
- Observe for evidence of haemorrhage at the wound site.
- Prevent licking/interference by applying appropriate bandage or Elizabethan collar.
- Reapply appropriate dressing material at recommended periods to enhance wound healing.

Bandages

An animal is likely to interfere with a bandage if it has become uncomfortable or too tight. In the event of patient interference, the VN should always check the bandage first. As a guide, one should just be able to insert a finger into the proximal and distal ends of the bandage. Boredom is another cause of interference so toys and time spent grooming the patient can help to remedy the problem. The bandage should be checked frequently and changed if it becomes soiled, wet or loose. Limb bandages should be applied to the entire limb; partly covering a limb may cause swelling distal to the bandage. If the toes are exposed, they should be assessed to ensure adequate blood flow to the area. An Elizabethan collar can be applied if necessary to prevent interference. When taking the animal outside, the lower part of the bandage should be covered with a protective covering. An empty drip bag with the end cut off provides a sturdy cover, which can be reused. The bag should always be removed as soon as the patient is returned to the kennel. Pressure bandages are removed after 12 hours of application.

Wound drains

Potential complications include wound breakdown, ascending infection, blockage and local tissue irritation. The VN must carry out effective hygiene procedures during the nursing of these patients and observe the animal closely for evidence of infection.

- Ensure that the drain is sutured securely to the skin.
- Clean the drain entry and exit site and surrounding skin daily with an antiseptic solution.
- Cover the drain entry sites with sterile keyhole dressings.
- Apply white soft paraffin around and below the drain exit site to prevent skin excoriation.

- Bandage area with highly absorbent dressing such as cotton wool to soak up secretions and secure in place using stockinette-type bandage.
- Prevent self-trauma by applying an Elizabethan collar.

Abdominal surgery

A serious complication of abdominal surgery is secondary peritonitis, which can lead to serious systemic illness and unless treated aggressively can be fatal. The onset of peritonitis is usually acute, occurring several hours to a few days following the surgery. Clinical signs include pyrexia (although if already shocked, the patient may be hypothermic), tachycardia, dehydration, abdominal pain and signs associated with hypodynamic septic shock. Close monitoring of the patient is essential in the initial postoperative period so early detection and treatment can provide the best possible prognosis.

- Observe the patient for signs of nausea, vomiting and abdominal pain.
- Maintain intravenous fluid therapy until the patient is able to meet its fluid requirements orally.
- Offer small amounts of water by mouth after the anaesthetic recovery, carefully observing the patient.
- After the initial food and water withholding time, encourage the patient to eat and drink to aid a return to normal gut peristalsis and electrolyte and nutrient assimilation.
- Gradually return to the patient's normal diet and feeding quantities over the next few days.

Aural surgery

The most frequently performed surgery to the ear includes drainage of aural haematomas, lateral wall resections and ear canal ablations. Following most aural surgery, the pinna of the affected ear is bandaged over the head to prevent excessive movements and head shaking (see Chapter 19). Facial nerve paralysis is a complication following canal surgery and may cause drooping of the eyelids and lips and dysphagia. These injuries usually resolve within a number of weeks, but the patient may require additional nursing care in the meantime.

- Check the head bandage frequently to ensure that it has not slipped or become too tight.
- Change bandages and dressing every 1–3 days.
- Clean the wound area gently using a mild povidone–iodine solution to remove crusts, discharge and haemorrhage.
- Protect wound and bandage from self-trauma using an Elizabethan collar.
- Observe the patient for evidence of facial nerve paralysis and apply an eye lubricant if required and as prescribed by the veterinary surgeon.
- Observe patient eating and assist if necessary.

Caesarean section (C-section, hysterotomy)

Caesarean section is the delivery of fetuses via a celiotomy and further incision into the uterus. These patients are frequently seen late at night or in the early hours of the morning. Some dams will not accept the young after a Caesarean. This may be overcome by waiting until she is recovered adequately from anaesthesia before placing them in the nest.

- Avoid disturbing the dam but quietly and frequently observe the dam until she is sufficiently co-ordinated not to damage the young and all of the pups or kittens are suckling.
- If possible, place the mother and offspring in a warm isolated area away from other noisy animals and human activity.

Care of the neonate after delivery

The surgeon will remove the puppy from the uterine horns and want to pass it on quickly so that he or she can continue with the surgery. The VN holds a towel in both hands and allows the surgeon to 'drop' the neonate into the open towel without contaminating the surgeon. The VN must then take the following steps quickly and efficiently:

(1) Using the roughness of the towel, break open the fetal membranes from around the neonate's head and body.
(2) Wipe the mouth and nose area with the towel to remove most of the fluid.
(3) Check to ensure a heart rate. If present continue steps 4–12. If absent, follow resuscitation procedure.*
(4) Use a cotton bud to gently prize open the mouth and use it to wipe away the fluid inside the mouth – go in as far as pharynx area.
(5) Use the towel to roughly rub the neonate to stimulate respiration.
(6) Respiration should now occur. Usually, a sharp, short inhalation is followed by short period of apnoea – keep rubbing neonate to stimulate (remember, they will still have the cardiorespiratory depressant effects of the anaesthetic agents).
(7) By now, the surgeon is probably ready to pass on another neonate so follow the above procedure.
(8) Place the first neonate into a box or large litter tray with hot water bottle/heated corn pad and clean bedding and keep checking it whilst sorting out the next neonate.
(9) Place new neonates next to the one before and at every opportunity, keep rubbing them to help stimulate them, help keep them warm and dry off their fur.
(10) Keep following the procedure shown above until all of the litter has been delivered.
(11) If there are more people around, e.g. the owner, get them to rub the neonates with a towel. By now they should be squeaking quite loudly.
(12) When all the neonates are delivered safely, tie off the umbilicus with gut suture material about 1 cm from the abdominal wall and cut just distal to the ligature.

*Resuscitation is covered in Chapter 40.

Cryosurgery

The postoperative changes that occur in the cryosurgical patient are quite profound and can distress an unprepared owner. The whole point of cryosurgery is to destroy tissues by freezing. This of course results in tissue necrosis and sloughing, which is most often the most disturbing aspect of cryosurgery visually. The frozen tissues undergo necrosis and a scab forms to protect the underlying healing tissues. This usually sloughs off in about 10 days to reveal a granulating wound.

- Apply a dressing and, if possible, a pressure bandage over the area. Swelling and haemorrhage are common within hours of treatment but should resolve within 48 hours.
- Clean the area daily with a mild povidone–iodine solution. If freezing involves mucous membranes or if the patient interferes with the site, the scab becomes moist and may smell offensive.
- Prevent self-trauma by fitting an Elizabethan collar.
- If the animal becomes anorexic, hand feed highly palatable food.

Gastric dilatation

Gastric dilatation–volvulus–torsion is a medical and surgical emergency and the immediate first aid measures are discussed in Section 8, Chapter 40. Potential complications in the patient recovering postoperatively from surgery to correct the condition include hypovolaemic shock, septic–endotoxic shock, arrhythmias and abdominal pain. Close observation is essential to detect any such complications early and prevent the situation becoming life threatening.

- Check core body temperature and treat hypothermia.
- Maintain fluid and electrolyte balance and ensure appropriate fluids are being administered at the suitable rate. Rates of up to 90 ml/kg/hr may be required during the initial therapy period.
- Observe for cardiac arrhythmias using electrocardiogram (ECG).
- Measure and record packed cell volume (PCV), sodium and potassium levels frequently (hypokalaemia is the most common electrolyte imbalance).
- Monitor urine output.
- Withhold oral food and water for 24 hours postoperatively.
- Start feeding after this period with small amounts of bland, liquidised food and watch patient following feeding for regurgitation.
- Raise the food bowl to help prevent aerophagia.

Gastrointestinal surgery

Surgery to the gastrointestinal tract is a fairly commonly performed procedure in small animal practice, frequently for the removal of a foreign body. Postoperatively, leakage from the gastric or intestinal incision can occur and peritonitis result. You must be fully aware of the clinical signs of septic shock so that it can be picked up early and treatment instigated. In addition to the

nursing care identified in the abdominal surgery care plan the following should be done:

- observe the patient for evidence of septic shock for the entire hospitalisation period;
- offer small amounts of water after the anaesthetic recovery;
- if water is retained, small quantities of food may be offered 12–24 hours following surgery. Food should be bland, for example, rice, boiled chicken or fish.

Ophthalmic surgery

Depending on the surgery involved it may be necessary to administer a number of different topical eye treatments. It is important that at least 5 minutes are left between administration of the different drops to allow absorption. Ophthalmic procedures range from eyelid surgery to intraocular surgery.

- Fit an Elizabethan collar before the patient recovers from the anaesthetic as eye surgery can be painful and these animals will often rub their eyes with their paws during the anaesthetic recovery period.
- Approach visually impaired animals slowly and talk to them gently so as not to startle them.
- Carry blind or poorly sighted animals outside rather than taking them on the lead, but if they are too heavy, lead them around obstacles and keep the leash short so they remain close to your side.
- Make up eye drop administration chart if topical applications are being administered frequently and record all treatment times.
- Carefully clean away discharges and crusts from around the eye using moistened cotton wool balls with eyewash or other suitable isotonic solution. Avoid touching or rubbing the eyeball itself.
- Following intraocular surgery, position the patient with the head higher than the body during the recovery period to help reduce the risk of retinal separation.

Oral/dental surgery

Surgery to the oral cavity includes repair of lacerated tongue, tumour removal, salivary mucocele, maxilla/mandible-ectomy and tooth extraction. After any oral procedure, there is a risk of inhalation of blood and/or mucus and the VN should check for airway obstruction frequently.

- Ensure that gauze sponges and mouth packs have been removed from the pharynx.
- Open mouth periodically to check for haemorrhage.
- Keep mouth clear of blood and saliva using swabs or suction.
- Observe patient closely for evidence of coughing or choking.
- Raise the patient's body and lower their head so that fluids drain out of the mouth.

- Offer water when the patient has regained consciousness but withhold food for the first 24 hours.
- After 24 hours, provide soft, moist food and hand feed if necessary.
- If the animal tries to scratch its face, fit an Elizabethan collar or bandage the feet if the collar interferes with the surgical area.

Orthopaedic surgery

Osteomyelitis is one of the most serious risks associated with orthopaedic surgery and therefore it is essential that the VN pays close attention to and records vital signs. The veterinary surgeon must be informed of any pain, swelling or discharging sinuses so that immediate action in the form of radiographs, bacteriology and sensitivity analysis and introduction of antibiotics can be initiated.

- Immediately following surgery, obtain core body temperature and treat hypothermia.
- Observe patient closely for signs of pain and inform veterinary surgeon as necessary.
- Keep the surgical wound clean and apply sterile dressing over incisional wound. Check, clean and change dressing daily.
- Ensure that all dressings and bandages are kept clean and dry and examine frequently to ensure that circulation is not compromised.
- Cover limb bandages and casts when taking the animal outside for exercise with a protective covering. An empty fluid bag with the bottom cut off makes a useful and tough cover. Always remove covering as soon as the animal is returned to the kennel.
- Support and assist patients with multiple fractures during exercise using a towel sling.
- Fit an Elizabethan collar if the patient interferes with the bandage or cast.
- Initiate physiotherapy if indicated but do not perform treatment over newly repaired fracture site.
- Consider high-protein diet following surgery and high calorie requirement of patient.

Ovariohysterectomy/pyometra

Ovariohysterectomy is one of the most commonly performed surgical procedures in many small animal clinics. With such a routine procedure it is easy to become blasé about the postoperative care of these animals and provide inadequate monitoring of the animal. One should remember, however, that haemorrhage is the most common cause of death following this type of surgery. Following ovariohysterectomy for pyometra, there is the additional complication of septic shock and the VN must be fully familiar with the associated clinical signs.

Extra attention, in addition to nursing care should be given to any patient following abdominal surgery.

- Monitor the patient closely for evidence of haemorrhage and hypovolaemic shock. Check for haemorrhage from the abdominal incision, abdominal swelling, pale mucous membranes, depression and weak, rapid pulse.
- Monitor closely for evidence of septic shock, which includes the typical signs of hypovolaemic shock such as tachycardia, tachypnoea, but instead the patient may show an elevated core body temperature and brick-red mucous membranes.

Spinal surgery

These patients require intensive nursing care. Spinal surgery can take several hours and hypothermia is common postoperatively as a result.

- Immediately following surgery, obtain core temperature and treat hypothermia as necessary. Remember that these animals may be physically unable to move away from heat sources so ensure burns do not occur from heat pads or lamps.
- Closely observe the patient for evidence of pain and refer to veterinary surgeon (opioids are most commonly used for the first 24 hours).
- Confine the animal to a small area to prevent too much movement.
- Provide lots of padding such as water beds, thick foam mattresses and Vetbed style bedding.
- Prevent skin contact with urine and faeces by using a non-retentive bedding material such as Vetbed. Application of white soft paraffin is also useful.
- Note the patient's ability to urinate and defecate and manually express or catheterise the bladder to prevent overflow or urine retention.
- Check the incisional area for evidence of swelling or haemorrhage.
- Apply ice packs and pressure wraps if necessary.
- Turn the patient 4-hourly to help prevent hypostatic pneumonia.
- Prevent decubital ulcers by protecting bony prominences.
- Perform a neurological examination daily.
- Provide physiotherapy three to four times daily to maintain muscle tone.
- Assist paraplegic dogs to walk using slings or a towel under the abdomen.

Thoracic surgery

As with all lengthy surgical procedures, hypothermia is extremely common postoperatively. In this case, a return to normal body temperature is particularly important because hypothermia and shivering increase oxygen consumption and decrease ventilatory capacity.

- Provide a quiet, stress-free environment so as not to aggravate existing respiratory insufficiency.
- During recovery, place patient in lateral recumbency with the side of the thoracotomy uppermost (if lateral thoracotomy incision performed).
- Observe the patient closely for signs of respiratory distress and evidence of postoperative pain (pain from a thoracotomy wound may prevent normal respiratory excursions and worsen ventilatory efforts).

- If available, measure tidal volume (should be no less than 10 ml/kg) using a Wright's respirometer and check oxygenation with a pulse oximeter.
- Thoracic bandages help seal the thoracotomy wound and reduce emphysema around the incision, but it is imperative that it is not too tight and restricting ventilation.
- Position patient in sternal recumbency as soon as possible to allow both lungs to expand and minimise congestion.
- Check thoracic drains frequently and record quantities of fluid/haemorrhage/air present in the chest bottle.
- Ensure drainage bottles are placed and maintained at least 1 m below the patient, providing a suction pressure of 5–10 cmH_2O.
- Remove drainage tubes as soon as possible; as a general guide, the tube can be removed if less than 100 ml of fluid is being drawn off over a 24-hour period.
- Provide supplementary oxygen therapy if necessary.

Urinary tract

Surgery includes nephrectomy, ureteric ectopia correction, cystotomy and urethrostomy. Following surgery to the urinary tract, temporary urinary obstruction may occur due to swelling postoperatively.

- Monitor urine output. Measure accurately if an indwelling catheter is placed or estimate the quantity if not.
- Observe patient closely for evidence of tenesmus, haematuria and pain on urination.
- Prevent urine scalding from overflow or leakage by applying a barrier cream such as white soft paraffin.
- Prevent self-trauma by fitting an Elizabethan collar.
- Maintain urinary catheters and collection bag systems. (Refer to care plans for general conditions.)
- Provide plenty of opportunities for the patient to urinate by taking outside frequently or providing a clean litter tray.

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SECTION 2

MANAGING THE HOSPITAL ENVIRONMENT

INTRODUCTION

This section covers the important points regarding running a veterinary practice. The main focus is how to minimise risk in the practice to both staff and patients. Health and safety is covered in detail, including how to undertake risk assessments in the practice as well as infection control and pharmacy management.

Chapter 6

Communication Skills in the Workplace

Communication and interpersonal skills are essential in veterinary nursing. In order for the team to work well together information must be exchanged or imparted to the members of the team. To a certain extent VNs communicate to their patients in order to decrease their stress of being in a strange environment. Animals easily pick up on cues from people. A VN should perhaps consider the animal and owner as the patient with whom she or he needs to communicate. Ensuring owner compliance means VNs need to ensure owners understand the treatment and care their pets require.

There are many ways in which communication can take place, however these are principally categorised into verbal and non-verbal (Table 6.1). Verbal communication suggests that the communication is purely in the form of language. When speaking on the telephone what we say is important, but the tone and pitch of a person's voice are also important and can alter the meaning. In face-to-face encounters there is a lot of non-verbal communication, which includes body language and facial expressions. This reinforces the message and VNs therefore need to ensure telephone communication is much clearer to be effective.

The type of language used is important to ensure an understanding between the 'giver' and the 'receiver'. The complexities of language mean that, although the receiver speaks the same language, he or she may be confused by everyday speech, including slang and non-literal meanings, and, in veterinary practice, the use of medical terminology.

There are several factors which can affect communication. When dealing with clients with sensory disabilities we often change the way we communicate (speaking loudly and slowly) which may not always be appropriate. It is important to find out from the client what suits them. For hearing loss clients one should find out how they communicate. Do they lip-read? Are they wearing a hearing aid? Would they prefer the information is written down? With sight loss clients, it is important to be sensitive to what they can do for themselves – do not grab their arm and march them into the consulting room! The environment can be obstructive to good communication, especially the noise in a veterinary practice reception area. The presence of the animal can affect how much attention the client is paying to the VN. It is known that, in order to ensure the client understands the aftercare requirements for their pet, the information should be imparted before the pet is reunited with the owner.

VNs will invariably come across a 'difficult' client. They may start shouting or even become physically abusive. It is important that the VN remains calm

Table 6.1 Forms of communication.

Mode	Advantages	Disadvantages
Verbal (face-to-face)	Appearance – first impression Facial expression/body language Tone of voice Control emotions Learn to read the body language of others Fast and direct Allows instant feedback	No record No control over content Verbal and physical abuse!
Telephone	Facial expression Tone of voice Control emotions No physical abuse!	Immediate action required – cannot leave the phone ringing Verbal abuse!
Written (letters, notices, memos, messages)	Accurate Clear and concise Spelling and grammar important Content control Clear and concise Usually unemotional	No instant feedback No physical or verbal abuse!

at all times and does not react. Pets play an important part in peoples' lives and owners naturally become upset or angry if they are worried about the welfare of their animal.

The following guidelines may help.

- Remain calm – do not react.
- People who want to make a scene like to have an audience so remove them to another place out of the waiting room.
- Let them explain – show you are listening and understand their problem.
- If you cannot help, refer them to a more senior member of staff.
- Do not take sides or place blame.
- Do not admit to anything unless the facts are clear.
- Using phrases such as 'I understand how you feel' may help.
- Someone who wants to have a shouting match finds it very difficult if the other person remains calm and doesn't react. They soon calm down.
- Do not take personal offence – or you will react!

Some practices install panic alarms in reception and consulting rooms. It usually depends on the area the practice is situated in. Staff should be aware of their safety at all times especially when working night duties. Two people should be present in the practice during out-of-hours duties although there is not a legal requirement for this. This ensures safety when dealing with the animals and also if there are unwanted callers late at night.

The style of communication can be equally important and this is where the pitch and tone can contribute to the overall message being imparted. Different styles are often described as:

- passive;
- passive–aggressive;
- aggressive;
- assertive.

People may utilise these styles in different circumstances and, in fact, effective communication means we should be using all styles.

QUESTIONING TECHNIQUES

The types of questions asked are important when trying to gain information particularly about the patient.

Closed questions tend to receive very short or one word answers, usually 'yes' or 'no'. For example we might want to find out how much exercise the dog gets. 'Do you take Ben for walks?' will probably initiate the answer 'yes'. This could mean 5 minutes in the morning or 3 hours a day. You may get the answer 'no', however the dog runs around the fields herding sheep all day.

Open questions are designed to obtain more information, encouraging the client to give a fuller description or explanation. For the above example the question might be, 'Tell me about Ben's daily exercise regime.'

Of course, closed questions can be useful in confirming or concluding conversations. Further examples have been previously discussed in Chapter 2.

LEARNING COMMUNICATION SKILLS

The importance of communication skills in veterinary practice has been recognised with communication skills being studied in veterinary schools. Good communication leads to a more satisfied workforce and an increase in client compliance. The Calgary–Cambridge model was developed in 2002 based on other models but with the focus being more veterinary specific. It is a useful framework for consultations and can be used for veterinary surgeons and nurses.

The Calgary–Cambridge model has seven major components, listed below, each of which has several subcomponents, amounting to approximately 70 in total:

- greeting;
- rapport building;
- history taking;
- clinical examination;
- explanation;
- treatment planning;
- closure.

The framework helps to guide the consultation and advises on the types of questions to be used whilst gaining the most information from the client. Students are initially exposed to the framework in the early years of the

course and the focus is more on communicating effectively rather than the clinical knowledge required. Further information may be found on the National Unit for Advancement of Veterinary Communication Skills website (http://www.nuvacs.co.uk).

TEAMWORK

Good teamwork is essential to creating a successful practice. This is not always easy as getting a number of personality styles working towards a common outcome can often result in clashes. Meredith Belbin (1981) described eight different team roles which complement each other.

- The coordinator – focuses on objectives, often leading the team, assertive.
- The shaper – will get things done, works well in stress, spurs the team into action but less tolerant than the coordinator.
- The plant – the ideas person (intelligent, creative) can be eccentric and introverted but will voice ideas.
- The resources investigator – extrovert member of the team with good communication skills, keen to network. Will have ideas but does not always follow them through.
- The monitor evaluator – a steady person who is useful at decision making as they can critically evaluate the situation in a dispassionate way.
- The company worker – organised, practical, disciplined. Will turn ideas into projects and get them done. Methodical hard worker.
- The team worker – keeps the team together diffusing tension and promoting team spirit.
- The completer finisher – similar to the company worker, will ensure the projects are completed. Conscientious and self-disciplined but will be concerned over detail. This can help prevent mistakes.

It can be important to establish the different roles of staff in order to appreciate different personality styles and how they interact with each other.

REFERENCE

Belbin, M. (1981) *Management Teams*. London: Heinemann.

Chapter 7

Record Keeping

Thorough and accurate records are essential. From a legal point of view certain records must be kept by the practice. The Royal College of Veterinary Surgeons (RCVS) recommend that the records in Table 7.1 are kept for a minimum of 6 years. The Veterinary Defence Society recommends that papers relevant to a disputed case are kept for 6 years and 364 days.

WHAT IS THE DATA PROTECTION ACT?

The Data Protection Act 1998 affects anyone who keeps and processes information about living individuals. The Data Protection Register lists all the people or organisations (known as data controllers) who keep such information. Any member of the public can consult this register, and each entry lists the name, address and the type of work carried out by the data controller, including their reason for keeping personal information. There are eight principles within the Act indicating that the information kept is relevant, accurate and must only be disclosed and used for the purposes registered. For a veterinary practice this usually means for mail shots, but may also be used for criminal proceedings or insurance purposes. Case records including radiographic films are the property of, and should be retained by, veterinary surgeons in the interests of animal welfare and for their own protection. However, if a client has specifically paid for them they are legally entitled to them.

Table 7.1 Records to be kept.

Client	Practice
Case records Radiographs	VAT records (6 years)
	PAYE records (at least 3 years after income tax year to which earnings relate)
Electrocardiograms (ECGs)	Invoices
Laboratory reports	Service contracts (these are usually annual)
Consent forms (anaesthesia/euthanasia)	Accident records (3 years)

CLINICAL RECORDS

Changes in a case can only be properly identified when they are written down and compared with previous recordings. Records also enable other members of the practice to become familiar with the case by perusal of the record sheet. Ensuring that care plans and instructions on the nursing interventions are explicit enables all the team members to continue the appropriate level of care required for the patient. The 'tick-box' case record sheet on the front of the kennel might help to reduce paperwork; however, simply ticking a box to show an animal has defecated does not provide information about how much and the consistency. This can also apply when monitoring food intake of an inappetent animal. A 'tick' can represent a mouthful or a bowlful.

Chapter 8

Minimising Risk in the Practice

HAZARDS AND RISKS

It is essential that the veterinary team is aware of the risks within a veterinary practice and the procedures and protocols necessary to try to reduce the risks associated with veterinary healthcare. The Health and Safety Executive (HSE) defines a hazard as '*anything to cause harm*'. The risk is the '*likelihood that something will cause harm*', usually described as low, medium or high risk.

The risk depends on the following:

- the hazard presented;
- how it is used;
- how exposure to it is controlled;
- how much the worker is exposed to and for how long;
- the work being done.

Regular risk assessments should be carried out to identify and control workplace hazards.

WHAT IS RISK ASSESSMENT?

A risk assessment is a careful examination of activities and equipment used which may cause harm to people in that environment (the veterinary practice). This would include employees, students on placements, clients and anybody entering the premises. A risk assessment helps to identify whether enough precautions have been taken or whether more are necessary to prevent harm. Employers are legally required to assess the risks in the workplace so that they put in place a plan to control them.

The risk assessment process involves five steps (described by the HSE).

(1) Identify the hazards.
(2) Identify who may be affected and how.
(3) Evaluate the risks and decide on precautions.
(4) Record findings and implement them.
(5) Review assessment and update if necessary.

Identify the hazards

Hazards that could result in significant harm with the veterinary practice are:

- slipping on wet floors;
- tripping over equipment or stock (bags of pet food etc.);
- chemicals used in the practice (disinfectants, medicines, radiographic chemicals and laboratory chemicals);
- electrical faults (especially equipment used);
- fumes (disinfectants and other chemicals used, anaesthetic agents);
- manual handling (animals, equipment and stock);
- noise;
- ionising radiation;
- poor lighting;
- extreme environmental temperatures;
- equipment (inadequate maintenance or training in the use of);
- animal bites and scratches;
- animal disease (zoonoses);
- stress.

Identification of personnel affected

Personnel who may be affected are:

- employees in the workplace (veterinary surgeons, veterinary nurses, students, patient care assistants, receptionists, cleaners etc.);
- members of the public (clients, contractors, visitors to the practice).

Inadequate training and lack of warning signs would contribute to the potential hazards identified.

Risk evaluation

Each hazard is assessed as to how likely it would be that it would cause harm. The risk is then classified as high, medium or low. The risk of being harmed by a very hazardous substance can be low if proper precautions are taken.

Documentation

Records of the risk assessment, including the precautions that should be taken, are filed in an accessible place where they may be consulted, reviewed and necessary changes made.

The veterinary nurse should be aware of key legislation involved in maintaining a safe environment, including:

- Health and Safety at Work Act 1974;
- Management of Health and Safety at Work Regulations 1999;

- Control of Substances Hazardous to Health (COSHH) Regulations 2002;
- Manual Handling Operations Regulations 1992;
- Ionising Radiation Regulations 1999;
- Reporting of Injuries, Diseases And Dangerous Occurrences Regulations (RIDDOR) 1995;
- Electricity at Work Regulations 1989;
- First Aid at Work Regulations 1981;
- Control of Pollution Act 1974;
- Collection and Disposal of Waste Regulations 1988;
- Environmental Protection Act 1990.

Review

Practices rarely stay the same; premises are altered, new equipment is obtained. It is important that the documentation is reviewed regularly, at least on an annual basis.

COMMON RISKS IN THE VETERINARY PRACTICE

Members of the veterinary team can be exposed to a range of hazards on a daily basis. A VN should be aware of these and ensure appropriate control measures are put in place to reduce the level of risk. Commonly encountered hazards include:

- manual handling;
- bites and scratches;
- zoonoses and infectious diseases;
- hazardous chemicals;
- clinical waste (chemicals, biological agents and sharps);
- ionising radiation;
- stress.

Pregnant employees

It is essential that hazards are identified which might affect the pregnant employee and that the level of risk is assessed. The pregnant employee may need additional supervision and advice. Both the mother and foetus may require protection from a potentially harmful external environment. Some risks are very relevant in the early stages of pregnancy when a woman may not realise she is pregnant. Employees who become or plan to become pregnant should be encouraged to make themselves known to the employer or relevant occupational health department as soon as possible, so that personal counselling can be initiated, and the appropriate advice given both to the individual and to management. Women of child-bearing capacity should be informed wherever an assessment reveals a risk and reassured that the new and expectant mother will not be exposed to any harmful risk. Specific advice regarding pregnancy is given under each section.

Risk management

All recent legislation relating to risks in the workplace requires the identification and assessment of risk and the introduction of measures that are reasonably practical to control them. For certain practices, such as manual handling, it is impossible to set clear limits beyond which risk is unacceptable. In other instances, such as exposure to hazardous substances, limits are set for pregnancy (e.g. ionising radiation) or for women of child-bearing capacity.

Employer's duties

Under the new regulations (Management of Health and Safety at Work Regulations 1999), employers are required to assess the health and safety risks to new and expectant mothers at work and take appropriate measures to protect them. Managers are required to carry out this assessment for any employee who is:

- pregnant;
- has given birth within the previous 6 months;
- breast feeding.

Health and safety risks include those to the unborn child or the child of a woman who is breast feeding as well as those to the mother herself.

Lifting in pregnancy

All lifting is considered in legislation relating to manual handling of loads and no pregnant woman should be required to undertake any lifting that might lead to placental disruption.

Most healthy members of staff are capable of lifting normally during the first few months of a normal pregnancy. After 20 weeks, lifting becomes more difficult. As the pregnancy advances, the woman's centre of gravity is progressively altered. Climbing ladders or steps may pose an additional risk and should be prohibited after the 24th week of pregnancy. Where pregnancy is at special risk (multiple pregnancy, elderly primipara and possible placental insufficiency) the expectant mother should be redeployed to areas where little or no lifting is required. If redeployment is not possible the employee should be advised to take sick leave after discussion with her general practitioner. Special attention should be given to mothers delivered by Caesarean section. Their ability to return to lifting may be impaired because of the surgical incision. There is no evidence that nursing mothers are at greater risk of injury through manual handling.

Standing

Duties requiring standing for long periods of time should be avoided in pregnancy. Seating should be available as far as possible, particularly in the latter weeks of pregnancy. Assisting at lengthy operations in theatre should be avoided.

Lone working, overtime, shift work and night work

The HSE provides guidance to employers about assessing the risks of lone workers. The practice should decide on the policy of whether it is acceptable to have someone working alone in the practice. Veterinary practices that have patients hospitalised overnight must ensure there are adequate precautions and protocols to ensure the safety of the person checking on these patients at night. The unpredictability of animals highlights the potential dangers of the lone worker in the practice.

The European Working Directive gives guidance on the average number of working hours per week. It is important to recognise the problems associated with working long hours including stress, fatigue and effect on personal and family life.

Most pregnant women can work the usual working hours but provision of time to rest after lunch may be required. Occasionally and on medical advice, shorter working hours or flexi-time may be required to avoid tiring rush-hour travel. Pregnant staff should not be required to work overtime and should not be required to work altering shift patterns which necessitate working a late evening shift. Pregnancy does not preclude the pregnant employee from working nights. Where a pregnant woman provides a medical certificate indicating that, in the interests of her health or safety, she should not work at night, night work should be prohibited with either a transfer to daytime work or paid leave from work where transfer is not applicable or practicable.

MANUAL HANDLING

Manual Handling Operations Regulations (1992)

The veterinary team is at high risk from work-related musculoskeletal disorders (MSD) as a result of the type of work undertaken. MSDs are the most common type of occupational ill-health in the UK. Until recently, guidelines to weight limits were given. The approach has now changed and is based on an ergonomic assessment according to various factors such as:

- the weight being lifted;
- the type of posture required (bending, twisting etc.);
- whether the item can be held close to the body or whether it is being lifted higher than shoulder height;
- floor surface;
- distance carried;
- presence of obstacles;
- team involvement.

The HSE has produced the manual handling assessment chart (MAC) to assist employers with their manual handling assessments (*Manual handling assessment charts* (INDG383)). Control measures that need to be put into place include:

- avoid hazardous manual handling operations so far as reasonably practicable (use hoists, trolleys etc.);
- assess any hazardous manual handling operations that cannot be avoided;
- reduce the risk of injury so far as reasonably practicable.

Adequate training of all staff in manual handling is required by the Health and Safety at Work Act. The content of the training should include:

- manual handling risk factors and how injuries can occur;
- how to carry out safe manual handling, including good handling technique;
- appropriate systems of work for the individual's task and environment;
- correct use of mechanical aids;
- practical work to allow the trainer to identify and put right anything the trainee is not doing safely.

As with any type of training, records should be kept of staff who have undertaken training courses and the content of such a programme.

Team manual handling

There are additional risks involved when more than one person is involved in lifting. The weight may not be evenly distributed, especially on slopes or steps. As a guide it is recommended that the capability of two people lifting is two thirds the sum of the weights that could be lifted individually. Factors affecting the lift include:

- lack of space for the number of people lifting;
- not enough good hand holds (especially with animals);
- background level of noise interfering with good communication between the team members.

It is essential that *one person* plans and takes charge of the lifting. Problems occur when the instructions are unclear. For example, '*After three, lift*' – some people lift when 'three' is said , others a few seconds after resulting in uneven lifting. This may also mean that 'one' and 'two' are omitted not allowing enough preparation time for the lift. It may be better to say 'lift on three; one, two , three (lift)'. It must be agreed between the members of the team when the lift will occur. This also includes the placing of the load (or animal) on a surface.

BITES AND SCRATCHES

Injuries sustained from handling animals in the course of working are very common. This is due to the unpredictable nature of the animals being treated and also inadequate training. The prominent canine teeth of dogs and cats can produce deep penetrating wounds. Bites lead to inoculation of potential harmful bacteria harboured in the mouths of these animals. The pressure that

a large dog can exert when biting also leads to extensive damage to tissues. Animals which are inadequately restrained may struggle leading to deep scratches which are also likely to become infected.

Prevention

All staff should be appropriately trained in both handling animals and using restraint equipment correctly. Using restraint aids properly is essential to prevent injury to personnel and to the animal. Where the animal is known to be aggressive or is showing signs of aggression, muzzling and sedation should be considered for all procedures unless contraindicated by the patient's condition. Signs indicating the animal may be aggressive or nervous should be clearly displayed on the front of the kennel. Case records may be stamped with 'CARE' in red to warn anyone handling the patient. Where this protocol is employed in the practice, further information should be provided on why and when the animal may become aggressive (e.g. the dog is fine to handle but will be aggressive when injected.)

Treatment

The risk of infection is particularly high in puncture wounds, hand injuries, wounds requiring surgical debridement and wounds involving joints, tendons, ligaments and fractures. Several potentially pathogenic species of aerobic and anaerobic bacteria are present in the oral flora of cats and dogs. Common genera include *Staphylococcus*, *Streptococcus*, *Pasteurella* and *Bacteroides*. Cat scratches can often cause 'cat scratch fever' where a number of organisms may be implicated (mainly *Bartonella*) causing localised lymphadenopathy. Superficial scratches should be washed under fast flowing water prior to being dressed. Wounds should be irrigated thoroughly to remove dirt and bacteria, with at least 200 ml of normal saline solution under moderate pressure (using a 20 ml syringe without a needle) and a first aid dressing applied. Any bleeding should be controlled as for usual first aid treatment; animal bites seldom result in major bleeding unless a major blood vessel is involved. Wounds on the hands or lower extremities frequently become infected and should be left open (i.e. not sutured). Puncture wounds should be washed under fast flowing water and an adhesive dressing applied. All animal bites and scratches should be referred to a medical doctor; antibiotic cover may then be prescribed for at least 3–7 days. A total of five doses of tetanus vaccine at suitable intervals is thought to provide the necessary cover to provide lifelong immunity.

ZOONOSES AND INFECTIOUS DISEASES

Zoonotic diseases (Table 8.1) are diseases that may be transferred from animal to man. Employees should be provided with personal protective equipment and suitable hand washing facilities. Wherever possible, pregnant women should avoid exposure to infectious diseases, especially viral diseases which pose the greatest potential threat to the foetus.

Table 8.1 Zoonotic diseases.

External parasites
Scabies – *Sarcoptes scabei*
Cheyletiellosis – *Cheyletiella*
Lyme disease – *Borrelia burgdorferi* (bacteria) acquired by tick bites

Fungi
Aspergillosis – *Aspergillus* rarely
Ringworm – *Micropsorum* and *Trichophyton*

Internal parasites
Visceral and larval migrans, toxocariasis – *Toxocara canis* and *T. cati*
Cutaneous larval migrans, hookworm disease – *Ancylostoma caninum*
Dipylidiasis – *Dipylidium caninum*
Echinococcosis – *Echinococcus granulosus*

Protozoan
Toxoplasmosis – *Toxoplasma gondii* (cats)

Viral
Rabies – Rhabdovirus

Bacterial
Leptospirosis:
L. icterohaemorrhagiae (Weil's disease) (rats)
L. canicola (canicola fever) (dogs)
L. hardjo (dairy worker fever) (cattle)
Pasteurellosis (*Pasteurella* spp., usually *P. multocida* – bacterium)
Salmonellosis – *Salmonella* spp. (dogs, tortoises, uncooked meat)
Tuberculosis – *Mycobacterium bovis* (cattle – raw milk)
Campylobacterosis – *Campylobacter* spp. (raw meat and milk and dog faeces)
Psittacosis, ornithosis – *Chlamydia psittaci* (birds)
Brucellosis – *Brucella canis* (dog)
'Cat-scratch fever' – *Bartonella* bacteria has been implicated

Toxoplasmosis

This is a worldwide infection in mammals and birds. Human infection is common. Unless known to have antibodies to *T. gondii,* pregnant women should avoid cleaning litter pans and contact with stray cats. Good clinical practice must be observed at all times with special attention paid to hand washing.

Vaccinia and related pox viruses

Smallpox vaccination is not generally recommended. Those judged fit to receive smallpox vaccination (as in the past) would be assumed fit to work with this class of virus. Pregnancy remains an exclusion.

Chlamydia species

These can cause septicaemia and foetal death in pregnant women. Pregnant women should therefore not bottle feed newborn lambs nor should they be

involved in any stage of the lambing process, particularly in the last trimester of pregnancy.

HAZARDOUS CHEMICALS

Any chemical agent can be potentially harmful, some are higher risk than others. In the veterinary practice the VN will be exposed to:

- pharmaceutical agents;
- cleaning agents;
- developing and fixer chemicals;
- laboratory chemicals.

A risk assessment will be carried out on the normal use of the chemical under the Control of Substances Hazardous to Health (COSHH) Regulations.

Pharmaceutical agents

If common sense rules are followed, there should be little danger of hazard to the health and safety of employees. Drugs may enter the body in different ways:

- absorption across the skin, e.g. prostaglandins, insecticides, vasodilators;
- absorption across mucous membranes e.g. sprays, powders;
- accidental ingestion, e.g. aerosol or powder-contaminated food;
- inhalation, e.g. halothane, powders, aerosols;
- accidental injection, e.g. any injectable agents from unguarded needles.

Particularly hazardous drugs include the following.

- Etorphine – highly toxic following injection or exposure of skin or mucous membranes.
- Halothane – repeated inhalation may cause liver damage or damage to the foetus.
- Cytotoxic drugs – carcinogenic, mutagenic (damage genetic material), teratogenic (damages the unborn foetus), e.g. Vincristine™, Endoxana™.
- Prostaglandins – may cause asthma attacks, affect the cardiovascular system and cause uterine contractions. They should not be handled by women of child-bearing age.
- Antimicrobials:
  - griseofulvin – teratogenic;
  - penicillins and cephalosporins – may cause hypersensitivity/allergic reaction;
  - chloramphenicol – can cause fatal aplastic anaemia (new blood cells fail to develop) in humans.

Anaesthetic agents

Gaseous anaesthetic agents should be effectively scavenged. Monitoring devices are used to ensure effective scavenging is taking place.

Concern has been voiced that anaesthetic gases may have a variety of deleterious effects upon pregnancy, including an increase in the miscarriage rate amongst staff exposed to waste gases as well as fetotoxicity and teratogenicity. High levels of nitrous oxide may cause an increase in miscarriage through its effect on vitamin B12 metabolism. This effect is less likely in modern theatre suites employing scavenging systems when the gas is vented out into the environment where it is massively diluted. Regular measurements should be used to confirm the efficacy of these systems and for the reassurance of staff. There is little hard evidence that other anaesthetic agents may have an adverse effect on pregnancy.

Concentrations of anaesthetic gases should be monitored and must not exceed the following limits (according to the Occupational Exposures Standards):

- halothane 10 ppm;
- nitrous oxide 100 ppm;
- isoflurane 50 ppm.

Unfortunately if any anaesthetic gas can be smelt the levels are about ten times over the limit. Precautions are discussed in Section 5.

Cytotoxic drugs

There are no specific legal requirements on the handling of cytotoxic drugs in the UK. Personal protective equipment should be used at all times. Animal studies have shown that at high doses many of these substances are mutagenic and carcinogenic and even teratogenic. There is evidence of some increase in markers of genetic damage amongst nurses. As a precautionary measure, however, pregnant or possibly pregnant women or nursing mothers should not be involved in the preparation or administration of cytotoxic drugs. This applies to nurses, doctors, pharmacists and pharmacology technicians. Special precautions should be followed.

- Prepare and administer toxic drugs in well ventilated and low-traffic areas.
- Wear correct protective clothing and equipment (face mask with eye protection or face mask and goggles, double gloves, apron).
- Administer through intravenous catheter to ensure the drug enters the vein.
- Use incontinence sheets to prepare the drug on and administer, potential spills may be absorbed immediately.
- The incontinence sheets and equipment may then be disposed of in clinical waste bags. Double bag all waste.

Cleaning and disinfectant agents

Formaldehyde is used in pathology laboratories and in low pressure steam autoclaves. The use of formaldehyde is subject to environmental monitoring and a ceiling limit for the UK is set at 2 ppm, which is said to be a safe level to protect all employees from any carcinogenic or teratogenic effects.

Cleaning agents should not be mixed and instructions for use must be followed. Disinfectants should be made up by adding the disinfectant to water in a bucket to prevent inhalation of vapour.

IONISING RADIATION

This hazard is met working with x-ray equipment (fixed, mobile and dental), computerised tomography and sealed (usually therapy) and unsealed (usually diagnostic or laboratory) radioisotope sources. Work with ionising radiation is covered by the Ionising Radiation Regulations 1999. Locations of work such as controlled areas and work activities that involve the exposure of staff including potentially pregnant or pregnant workers to ionising radiation are explained in detail in the Local Rules and Systems of Work. Advice on these is best sought from the local Radiation Protection Supervisor or the Radiation Protection Adviser as indicated in the Local Rules. Further detail on taking precautions is discussed in Section 6.

NON-IONISING RADIATION

Ultrasound

There is no proven link between staff involved in ultrasound imaging and an unfavourable outcome to pregnancy.

Magnetic resonance imaging (MRI)

The powerful magnetic force can present problems for employees with metallic implants. Other problems can occur when metallic equipment is taken into the MRI room. These are described as 'missile effect' accidents. Care must be taken especially with non-MRI-compatible anaesthetic equipment and patient trolleys. Noise from the coils can also be a potential problem if employees are exposed for long periods of time. Ear plugs or MRI-compatible headphones should be used. It has not been shown that MRI has any effect on the foetus and the IRPA (International Radiation Protection Association) makes no special recommendations on employment in pregnancy.

Medical lasers

The use of lasers in medical practice is well controlled in order to avoid hazards to patients and staff, particularly from damage to the eye which may be due to faulty equipment, misdirected beams, inappropriate laser settings or reflected beams. Signs should be displayed on the outside of the room where a laser is being used. All personnel in the room should be wearing safety glasses. Pregnant staff are at no extra risk.

Display screen equipment

Due to the nature of the job it is unlikely that a veterinary nurse will spend long periods of time at a computer or other display screen equipment (DSE).

It is advised that a 5–10 minute break should be taken every hour when using DSE. There is no evidence of any harmful effects as a result of using DSE in pregnancy and no special recommendations are necessary.

CLINICAL WASTE (CHEMICALS, BIOLOGICAL AGENTS AND SHARPS)

All commercial establishments, including veterinary practices, are classed as *industrial users* with regard to disposal of waste materials. All waste generated from practices is industrial waste (rather than household or commercial waste). The following legislation is concerned with regulation of storage, transfer and eventual destruction of waste products:

- Controlled Waste Regulations 1992;
- Control of Pollution Act 1974;
- Environmental Protection Act 1990.

Generally, waste can be considered to be any article or substance which is scrap material and is broken, worn out, contaminated or spoiled or is effluent or other unwanted surplus arising from any process. Emissions to the atmosphere are subject to the Environmental Protection Act 1990 and discharges to sewers of trade effluents are controlled under a licensing system regulated by the sewage undertaker.

The control and disposal of particularly dangerous or hazardous substances, e.g. lead, asbestos, explosives, radioactive substances, is subject to specific legislation which, together with associated codes of practice, should be consulted when such wastes occur.

Clinical waste is defined by the Controlled Waste Regulations 1992 as

> any waste which consists wholly or partly of human or animal tissue, blood or any other body fluids, excretions, drugs or other pharmaceutical products, swabs or dressings, or syringes, needles or other sharp instruments, being waste which unless rendered safe may prove hazardous to any person coming into contact with it.

Clinical waste is further categorized by the Health Services Advisory Committee according to its potential risk to health and safety into groups A to E (Table 8.2). Clinical waste sacks should conform to BS 6642:1985. Sacks should be coloured opaque yellow with the words 'Clinical waste – For Incineration Only' clearly printed on the outside. They should be collected by the local authority or licensed contractor for incineration. Sharps containers should conform to BS 7320. They must be leak proof and puncture resistant. They must be coloured yellow and clearly marked with the words 'Danger Contaminated Sharps – To Be Incinerated'.

The British Veterinary Association has further advice on their website www.bva.co.uk.

Animal bedding

Bedding from animal cages is not classified as clinical waste. However, as the VN cannot be sure of the extent of contamination of veterinary bedding it

Table 8.2 Categories of clinical waste.

Group	Description of waste	Method of disposal
A	Animal tissues, organs and blood Cadavers Animal body fluids (e.g. urine, vomit, faeces) Products containing body tissues or fluid (e.g. soiled swabs and dressings)	Clinical waste sack
B	Discarded syringes, needles Small items of broken glass Other sharp instruments (e.g. surgical blades)	Place immediately into approved sharps container, seal when full. Collected by licensed contractor
C	Microbiological cultures Potentially infected waste from laboratory	Autoclave before disposal to ensure microbes are killed
D	Drugs and pharmaceutical and chemical waste	Bottles and vials contaminated with pharmaceutical products should be put into green rigid containers and collected by licensed contractor for incineration
E	Items used to dispose of urine, faeces and other bodily secretions which do not come under group A	Clinical waste bags

may be advisable for newspaper, cat litter and any other extraneous bedding to be treated as clinical waste.

Schedule 2 drugs

Controlled by the Misuse of Drugs Act, Schedule 2 drugs must only be disposed of in the presence of a Home Office Inspector. Records must be made in the Dangerous Drugs Register for the Home Office Inspector to sign.

Cadavers

Cadavers are considered clinical waste. They should be placed into heavy-duty body bags, sealed and frozen prior to collection by a licensed contractor for incineration. Owners who wish to take their pet home for burial should be made aware of requirements by the Department of Environment. They indicate that animals suffering from infectious or hazardous diseases should be dealt with by the veterinary practice and classed as clinical waste. Low-hazard deceased pets may be disposed of by the owner at home within the curtilage of their dwelling in their capacity as a private individual without breach of duty of care. It is normal to get the owner to sign a disclaimer form if they wish to dispose of their pet's body at home. It is important that it is buried deep enough to allow a layer of stones on top to prevent the digging up of the site by wild animals.

Chapter 9

Infection Control

Good hygiene and standard infection control precautions are crucial measures to protect veterinary patients and ensure safe clinical practice. The VN has a responsibility to ensure that he or she is aware of all the associated risks and has received appropriate training in hygiene and infection-control measures. It is essential that the necessary steps in protecting patients and helping prevent the spread of communicable diseases and nosocomial infections are taken. This will mean drawing up practice protocols in hygiene measures and informing and educating veterinary colleagues, new members of staff and students on work experience. The following is an example of a standard operating procedure (SOP) for cleanliness in the practice.

- All employees should take great care with their personal hygiene to avoid risk of zoonotic diseases (see COSHH risk assessments) and transferring infection between patients. Wash hands after handling each animal, animal products or drugs.
- Use protective clothing provided, i.e. disposable plastic aprons, white lab coats, gloves, masks, goggles and hats when necessary.
- The hospital should be kept clean at all times. Each person is responsible for ensuring that they clean up after themselves as they work.
- All floors must be kept free from litter, animal waste/blood, hair and dirt.
- Appropriate cleaning and disinfection should be carried out on a regular basis (as often as necessary in heavily used areas). Warn others of wet floors (use signs) if necessary. Doors and handles must also be cleaned on a regular basis.
- Consulting and operating tables, kennels, cat baskets and weighing scales must be cleaned and disinfected between patients.
- Operating theatres should be damp dusted before use and thoroughly cleaned and disinfected after use.
- Bacterial air filters must be changed as often as recommended.
- Eating and drinking are forbidden in all clinical areas of the hospital.
- Smoking is forbidden in all areas of the hospital.

The most usual means of spreading infection include:

- hands of staff and personnel;
- inanimate objects (e.g. clothing, bedding, kennels, brushes, food bowls, etc.);
- dust particles or droplet nuclei suspended in the atmosphere.

By ensuring that the following precautions are applied at all times the VN can help to significantly reduce the risk of infection:

- maintaining hand hygiene;
- use of protective clothing and equipment;
- correct and safe use and disposal of sharps;
- correct and safe disposal of clinical waste;
- decontamination of clinical equipment;
- maintaining environmental cleanliness and hygiene.

HOSPITAL-ACQUIRED INFECTIONS

Hospital-acquired infections (HAIs) or nosocomial infections are prevalent in human hospitals as well as veterinary hospitals and practices. These are diseases which the patient contracts whilst they are hospitalised. HAIs may be endogenous or exogenous.

- Endogenous infections are infections acquired from the patient's own normal flora.
- Exogenous infections are mainly acquired from infected patients or healthy carriers/medical equipment/personnel. Transmission of exogenous infections is mainly on the hands of medical personnel. Risk of infection increases if the animal undergoes invasive procedures, from intravenous catheters, urinary catheters and arthrocentesis to surgery. The bacteria which are implicated in the HAIs are often common bacteria which have become resistant to antibiotics.

Methicillin-resistant *Staphylococcus aureus*

Staphylococcus aureus is a bacterium which is widespread in the community. It is carried in the nostrils of about 30% of people and may be present in other sites such as the throat and the skin, particularly in sweaty areas, e.g. axilla and groin. It establishes itself readily on damaged or abnormal skin, e.g. wounds, ulcers etc. Animals commonly carry *Staphylococcus intermedius* rather than *S. aureus*, although they are still susceptible to *S. aureus*. Methicillin-resistant *Staphylococcus aureus* (MRSA) was first described in 1960 in the UK and has since been reported around the world. This particular strain of *S. aureus* became resistant to the antibiotic methicillin, hence the name MRSA. MRSA is often also resistant to several other commonly used antibiotics, particularly those taken orally.

In normal healthy people, carriage of MRSA is usually symptomless and is not associated with risks to their health, even if they are pregnant, or to the health of the people they live with. MRSA may be passed from person to person, by direct contact via hands or touching an affected surface. The commonest method of spread is by hand. Swabs of staff may be taken to identify carriers who may then be treated. During treatment, the individuals may be advised to avoid any invasive procedures with animal patients.

Personal hygiene

Personal hygiene is an essential part of infection control. Microbes can easily be transmitted via hands and clothing. In addition to maintaining a professional appearance, personal hygiene is important to good client relationships as well as representing the profession. Jewellery can act as a source of infection, including wrist watches. Animals can also get caught in earrings and necklaces when being closely restrained. Long hair should be tied back so that it does not become contaminated. Shoes should cover the toes and be substantial to prevent injury to feet if an animal walks on them or something is dropped on them.

Hand washing

Research in human nursing has shown that hand washing is the single most important procedure for preventing nosocomial infection as hands have been shown to be an important route of transmission of infection. Even wearing rings increases the number of microorganisms on the hands. The procedure is often not carried out in an effective or satisfactory manner. Several methods have been published describing a thorough routine (Fig. 9.1).

Protective clothing

Personal protective equipment (PPE) should be provided for staff in the practice. It is the employee's responsibility that it is worn. The veterinary nursing uniform is both protective clothing but also signifies qualification. Silver impregnated uniforms are now available to help with infection-control procedures, however disease (or parasites) may still be transmitted. It is good practice to change into the uniform upon arrival at work and change again when leaving. This will help prevent the spread of infection to animals outside of the practice (own pets). Wearing plastic aprons over the uniform and changing them frequently helps to prevent contamination of the uniform. Gloves should be worn to protect oneself from infection and also help to prevent the spread of infection via the hands. Beware of opening doors and cupboards after dealing with an animal when wearing gloves. Always wash hands before and after wearing gloves.

Other protective clothing includes plastic glasses or goggles, face masks and hats. These must be used to prevent contamination of mucous membranes (nose, eyes etc.) and to prevent contamination from hair.

MAINTAINING ENVIRONMENTAL CLEANLINESS AND HYGIENE

It is the VN's role to ensure effective disinfection of practice, kennels and equipment to prevent cross-infection and nosocomial infections. Patients suspected of, or known to have, a contagious/zoonotic disease should be isolated. Restricted access to these cases should reduce the risk of spread to other patients and staff. Personal hygiene must be carried out before handling each patient, their fluid administration lines, wounds and before giving any

Fig. 9.1 Hand washing procedure.

Remove hand and wrist jewellery (a plain wedding band may be worn)

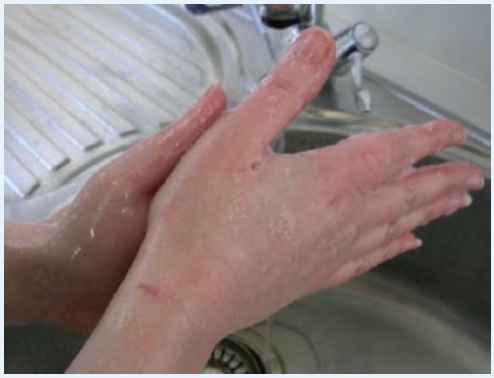
Turn on the taps to produce a steady flow of comfortable temperature of water

9.1a

Wet hands and wrists thoroughly (Fig. 9.1a)

Depress soap dispenser pump and apply 2–3 pumps of antimicrobial hand wash (e.g. chlorhexidine) on to your free hand (Fig. 9.1b)

9.1b



9.1c

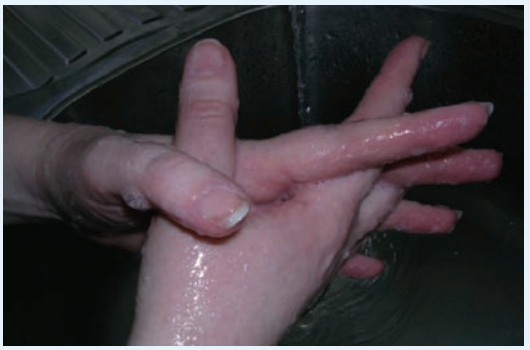
Step 1: Rub hands together – palm to palm (Fig. 9.1c)

Continued

9.1d

Step 2: Place the palm of the right hand over the back of the left hand and rub together (Fig. 9.1d)

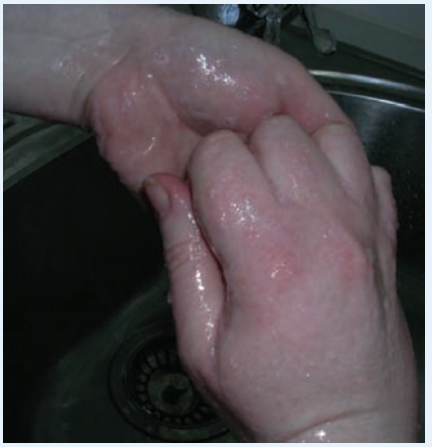
Repeat with left palm over back of the right hand



9.1e

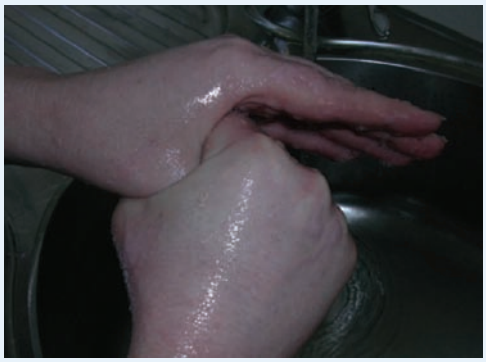
Step 3: Interlace the fingers of both hands and rub together (Fig. 9.1e)

Step 4: Interlock fingers of both hands – backs of fingers touching opposing palms – and rub together (Fig. 9.1f)



9.1f

Repeat on the other side, i.e. so left hand is uppermost



9.1g

Step 5: Clasp the left thumb in the right hand and rotationally rub together (Fig. 9.1g)

Repeat for the right thumb

Step 6: Using fingers of the right hand held together, place in the left palm and rub backwards and forwards (Fig. 9.1h)

9.1h

Repeat for the other hand

Step 7: This is an optional extra step (not all authorities advise)

Grasp wrist of left hand with right hand and rub rotationally (Fig. 9.1i)

9.1i

Repeat for the other wrist

Continued

Rinse both hands and wrists thoroughly under the running water

9.1j

Turn off taps with the elbows (Fig. 9.1j) (or use a paper towel if appropriate for the taps) (Fig. 9.1k)

9.1k

Dry hands with disposable paper towel

Dispose of used paper towel into foot-operated or open topped bin

injections etc. All faeces, urine, vomit, etc., must be cleaned up immediately and food must not be left around for too long as it will go off and attract flies. Although this deals mainly with the ward area, the principles apply elsewhere in the practice. Theatre cleaning is covered in Section 3.

Decontamination of clinical equipment

- Decontamination – a combination of processes which removes or destroys contamination so that infectious agents or other contaminants cannot reach a susceptible site in sufficient quantities to initiate infection or other harmful response.
- Cleaning – physical removal of organic matter and infectious agents.
- Disinfection – reduction in viable infectious agents.
- Sterilisation – rendering an object free from all viable infectious agents.

Correct decontamination of equipment ensures infections are not transmitted between patients. Many items are now disposable to prevent the possibility of transmission and it is essential that they are only used once. Diagnostic equipment and instruments used in the consulting rooms should be sterilised between each patient – this may mean using a cold chemical sterilisation process for convenience.

Cleaning accommodation and equipment

The protocol in Table 9.1 should be employed for cleaning accommodation. Equipment in the ward must also be cleaned and disinfected regularly to maintain effective hospital hygiene. Steps C to I listed in Table 9.1 may be used for general cleaning and disinfection of pieces of equipment listed below. In addition, some of these items may then require sterilisation using an appropriate method.

- Food bowls.
- Grooming brushes.
- Mops.

Table 9.1 Protocol for cleaning accommodation.

	Action	Rationale
A	Remove animal to secure outside run or temporary cage (must not be another animal's kennel)	Kennel cannot be cleaned effectively with animal inside and increases risk of escape
B	Remove bedding, newspaper, food bowls and toys	Bedding and bowls need washing, dispose of other bedding materials appropriately
C	Remove gross soiling (faeces etc.) with shovel or dustpan	Must be removed before attempting to clean. Dispose of appropriately
D	Clean with detergent solution	To clean away dirt and debris to prepare for effective disinfection – many disinfectants are inactivated by organic matter
E	Rinse with water to remove detergent	Many disinfectants produce noxious gases when mixed with detergent or become inactivated by these solutions
F	Apply appropriate disinfectant taking their recommended use and dilution rates into consideration	Some species of animal are sensitive to some types of disinfectant (e.g. cats and phenol)
G	Leave for recommended contact time	To ensure most effective destruction of pathogenic microorganisms
H	Rinse thoroughly	Strong odours may be offensive and/or irritant to some animals
I	Dry thoroughly	Or newspaper layer will become wet
J	Replace fresh bedding materials if necessary	Provide comfort and warmth
	Return animal to kennel if necessary	Ensure animal's security

- Shovels.
- Bedding.
- Bins.
- Kitchen utensils.

SELECTING APPROPRIATE CLEANING PRODUCTS

The following should be taken into consideration when selecting a particular antiseptic or disinfectant:

- effectiveness against particular organisms, e.g. parvovirus;
- effectiveness against a wide range of microorganisms (Gram-negative bacteria and bacterial spores, fungi and viruses which are more resistant than Gram-positive bacteria);
- toxic and irritant effects on operators and animals;
- effectiveness in the presence of a wide range of organic and inorganic materials;
- stability of product in storage or once made up;
- smell of the product;
- cost of the product;
- staining or corrosive effects on certain materials;
- contact time required;
- ease of use;
- Control of Substances Hazardous to Health (COSHH) Regulations and handling precautions required;
- effectiveness in different temperatures of water;
- possible toxic or noxious effects when mixed with other substances such as detergents.

Detergents

Detergents are primarily soap cleansing agents such as washing-up liquid and washing powder. They do not necessarily destroy microorganisms, although transient bacteria may be removed by thorough washing with such an agent and water. The main function of detergents in practice is to remove dirt, grease, body fluids and other organic materials in preparation for disinfection or sterilisation.

Antiseptic solutions and antimicrobial hand rubs

Antiseptic solutions and hand rubs are used primarily to destroy or inhibit the growth of microorganisms on the skin and mucous membranes. Many antiseptic solutions also contain a soap/detergent (surfactant) solution.

There are four main groups of antiseptics or skin disinfectants: chlorhexidine, iodophors, triclosan and alcohol. Refer to Chapter 16 for further information.

Disinfectants

Disinfectants remove or destroy microorganisms (although not always bacterial spores) in the environment. They must be used in accordance with the manufacturer's recommendations to ensure their effectiveness.

Ideal properties of antiseptics and disinfectants include:

- adequate destruction of harmful microorganisms on skin and surfaces;
- able to kill viruses;
- able to work in clean and dirty situations;
- must be safe for patients and staff;
- non-corrosive;
- must not stain;
- economical;
- stable for storage;
- pleasant odour;
- efficient and quick in action.

Unfortunately there is no one ideal solution which will do all this so it is important to select the most appropriate product for the situation. Changing the disinfectant that is used throughout the practice on a regular basis is good practice, helping to reduce the incidence of 'superbugs'. Also, when deciding on which disinfectant, the following factors should be considered.

- Inactivation by organic matter. Some disinfectants may become inactivated by the presence of organic matter such as urine, faeces, vomit, blood etc., therefore the kennel should be cleaned with a detergent and water first. Some newer disinfectants contain a detergent to help clean. Some rubber, cork or plastics may also affect the activity of the disinfectant.
- Staining of bedding. The disinfectant may stain bedding and other porous materials.
- Hard or soft water. You need less detergent in soft water and this is therefore more economical.
- Species sensitivity. Cats and phenols! Be careful with other species such as birds and reptiles.
- Strong odour. Particularly strong odours or fumes can cause ocular and nasal irritation both in humans and animals. Animal sense of smell is far more sensitive than humans.

Effectiveness of a disinfectant

The disinfectant will have been tested for its efficiency against specific microorganisms (Table 9.2). These results will also show the correct strength or dilution and contact time. The tests are the Rideal-Walker and Chick-Martin tests (only really useful for phenolic based disinfectants) or more recently the Kelsey-Sykes test. Groups of disinfectants are described in Table 9.3.

Table 9.2 Types of disinfectants which are more effective against certain microbes.

Disinfectants that have been found to be most effective against bacterial spores	Disinfectants known to be effective against viruses
Aldehydes Ethylene oxide Halogens Hypochlorites	Aldehydes NaDCC (sodium dichloroisocyanurates)

Table 9.3 Major groups of disinfectants.

Group	Qualities
Alcohol, e.g. isopropyl alcohol 70%, ethanol 70%	Effective against bacteria but not spores and some viruses Inactivated by organic material Highly flammable Expensive Skin irritant
Aldehyde, gluteraldehyde (Cidex™)	Effective against a wide range of bacteria, spores and viruses Not inactivated easily Slow acting Highly toxic causing skin and eye sensitivity and respiratory problems Activity is related to temperature
Peroxides, e.g. hydrogen peroxide, Virkon™, Oxykill™, hypochlorite	Wide range of activity Fast acting Ineffective with organic matter (although some newer disinfectants in this group are not affected) Low irritation and toxicity Virkon can cause metal corrosion

To maximise the efficiency of a disinfectant note the following points.

- Use at correct concentration:
  - too weak it is ineffective;
  - too strong is wasteful and possibly irritant.
- Do not mix with other detergents or chemicals.
- Use at correct temperature. They are usually more effective in warm water.
- Allow correct contact time.
- Rinse thoroughly after use to remove residues.
- Use freshly made up solutions.
- Some disinfectants are less efficient in hard water.

Points to remember when using disinfectants.

- New products come on to the market all the time. Keep up to date with new products and chemical compounds.
- VNs must always look after their own safety and hygiene and wear appropriate protective clothing. This ranges from basic gloves and apron

for all disinfectant products to wearing protective goggles and mask with others (aldehydes). Do not be lazy about this – effects may not be immediate but become evident years later.
- Always look after the safety of the patients – do not use chemicals that are irritant or toxic to them.
- Store disinfectants according to manufacturer's instructions and once made up use within recommended time.

RUNNING AN ISOLATION UNIT

The VN needs to have an understanding of how common infectious or contagious diseases are spread in order to reduce the spread of infection in the practice. Any animal that is suspected of having a contagious disease should be admitted into the isolation unit, away from any other animal. They should have their own outside runs, bedding and bowls. Personnel should wear specific clothing and carry out scrupulous hygiene procedures after handling any infectious animal. This is called barrier or isolation nursing.

Remember that contagious diseases can be transmitted by:

- direct contact: animal is in close contact with another or bites from fights;
- indirect contact: aerosol droplets, people, inanimate objects (fomites), e.g. food bowls, bedding and kennels.

Basic design of the isolation unit

The isolation unit should be a totally self-contained unit, containing all the bedding, feeding, monitoring and fluid therapy equipment required by each particular case. As with all kennelling, there should be an active ventilation system, which allows at least 6–12 air changes per hour. Heating systems must be thermostatically controlled to provide the required ambient temperature for each animal, bearing in mind that many isolated cases have the potential to become hypothermic easily. This is of particular importance to animals with severe diarrhoea, for which provision of blankets can be difficult. The walls and floors should be easy to clean and disinfect, ideally with a central drainage system to enable the whole room to be hosed down and disinfected. There should be as little clutter as possible. All equipment and bedding, etc. should be kept in closed cupboards and surfaces kept clear. It should be remembered that inanimate objects can transmit disease. Written protocols can help to ensure that barrier nursing is carried out effectively by everyone.

Isolation unit protocol

(1) Keep the patient's records and hospitalisation sheet on the outside of the room so that anyone can peruse them without having to enter the room.
(2) Place a sign on the front of the door to the unit to warn staff what the potential danger is, e.g. canine parvovirus, leptospirosis, etc.
(3) Wear shoe covers or specific footwear, and disposable gowns, gloves and mask when handling the patient or cleaning the kennel.

(4) Dispose of protective clothing into the clinical waste bins.
(5) Wash hands thoroughly in an antiseptic solution upon leaving the unit.
(6) Place a foot-bath (litter tray filled with disinfectant solution) just outside the door of the unit and instruct personnel to walk through it when leaving the isolation unit.
(7) Involve as few people as possible in the treatment of the patient (i.e. the attending veterinary surgeon and one nurse).
(8) Attend to the patient *after* treating any other inpatients and not *before* treating them. Isolation staff are not subsequently to attend to young or old animals or those who are immunosuppressed.
(9) Provide the patient with disposable bedding such as newspaper and incontinence sheets. Dispose of all bedding into clinical waste bins.
(10) Wash all used food bowls in detergent, and leave to soak in a disinfectant solution for appropriate length of time. Autoclave metal bowls before using for another animal.
(11) Do not allow the patient to come into contact with any other animal during its stay.
(12) Take isolated patient to a separate designated run or area to urinate and defecate.
(13) Clear away all urine, faeces, vomit etc. as soon as it is voided and use disinfectant as recommended by the manufacturers.

Chapter 10

Managing the Pharmacy

VNs have increasing responsibility within the pharmacy area of a veterinary practice. Additional qualifications which allow the nurse to dispense certain groups of drugs are now available. The level of knowledge that a VN has regarding drugs has meant a qualified VN can 'top-up' to become a Suitably Qualified Person (SQP) and be registered with the Animal Medicines Training Regulatory Authority (AMTRA). AMTRA is the professional body for SQPs, covering training, professional ethics and representation. SQPs can then legally prescribe and dispense a number of drugs, mainly worming medicines.

Veterinary drugs have previously been controlled by the Medicines Act 1968 and the Medicine Regulations 1994. In 2005 the Veterinary Medicines Regulations were introduced. The main changes were to the dispensing and prescribing groups. The regulations are updated annually and come into force every October. The Veterinary Medicine Directorate enforces the Veterinary Medicine Regulations.

The VN's role in managing medicines is varied and carries responsibility to:

- administer medications correctly;
- dispense drugs correctly;
- inform clients of side effects and withdrawal times;
- calculate dosages;
- recognise adverse reactions and drug incompatibilities and report them to the veterinary surgeon.

Prescribing medications is primarily the role of the veterinary surgeon. However an SQP can prescribe certain groups of drugs depending on their qualification. The following are the current distribution categories under the Veterinary Medicines Regulations.

- Prescription Only Medicine – Veterinarian (abbreviated to POM-V) may be prescribed and dispensed *only* by veterinary surgeons for animals *under their care* (or by a pharmacist on instruction of a veterinary prescription) (previously POM, and some P products).
- Controlled Drugs (CD) are a sub-category of POM-Vs. The rules for supply and storage of these drugs are even more stringent than for general POM-V medicines.
- Prescription Only Medicine – Veterinarian, Pharmacist, Suitably Qualified Person (abbreviated to POM-VPS). Prescribed by any one of the Registered

Qualified Persons and supplied by any one of them (previously PML, and some P products).
- Non-Food Animal – Veterinarian, Pharmacist, Suitably Qualified Person (abbreviated to NFA-VPS). Supplied by any one of the Registered Qualified Persons (previously PML and some P products).
- Authorised Veterinary Medicine – General Sales List (abbreviated to AVM-GSL). Supplied by any retailer (previously GSL products).

An SQP may supply products that have been authorised with a distribution category of POM-VPS, NFA-VPS or AVM-GSL.

A Registered Qualified Person (RQP) is defined as:

- a veterinary surgeon registered with the Royal College of Veterinary Surgeons;
- a pharmacist registered with the Royal Pharmaceutical Society of Great Britain or the Pharmaceutical Society of Northern Ireland;
- an SQP who is registered with AMTRA.

ADMINISTRATION OF MEDICATIONS

One of the responsibilities of the VN in the practice is the preparation of medications prescribed by the veterinary surgeon and ensuring that inpatients receive the correct dose at the correct time. By following the 'five rights' of drug administration, medication errors can be avoided.

(1) Right animal – which black cat?
(2) Right drug – what strength is the drug?
(3) Right dose – correct calculation?
(4) Right time – BID, Q6h?
(5) Right route – oral or topical?

It is also essential that the right documentation is completed accurately, i.e. prescription, dispensing label, Controlled Drug records.

Right animal

All patients should be easily identified. Using paper collars with animals' details on can help eliminate errors involving administration of medications to the wrong animal. Two black cats can look very similar!

Right drug

Check the label of the container with the patient's records. Ensure the correct strength and formulation. Ensure the drug has not expired. The person administering or dispensing the drug should be familiar with the action of the drug and any contraindications. Check the generic name, as one drug can have many trade names as it may be manufactured by several companies.

Right dose

The dose of a drug varies depending on the manufacturer. For this reason, the VN must always state the dose of a drug in units of mass, such as the milligrams or grams rather than the number of products or volume (millilitre, ml).

For example, writing 3 ml of xylazine (Rompun™) is unacceptable because it is available as concentrations of 20 mg/ml and 100 mg/ml. Therefore, 3 ml could contain either 60 mg of the drug or 300 mg. In many cases such an error could be fatal.

The calculation to find out an amount is:

$$\text{Dose prescribed (mg)} \div \text{Concentration (mg/ml)}$$

For example, a 20 kg dog needs 20 mg/kg of ampicillin. The ampicillin comes as a concentration of 100 mg/ml.

$$20 \times 20 = 400 \text{ mg}$$

$$400 \div 100 = 4 \text{ ml}$$

Right time

The time between drug dosages is often expressed using Latin abbreviations and it is essential that the VN is fully aware of them. Commonly used terms include:

- SID once daily (every 24 hrs);
- BID twice daily (every 12 hrs);
- TID three times daily (every 8 hrs);
- QID four times daily (every 6 hrs);
- Q4h every 4 hours;
- o.d. every day;
- e.o.d. every 2 days (every other day);
- Prn as needed.

The dosage interval may be adjusted to suit either the client or hospital staffing times and still give the required amount over a 24-hour period. However, drug manufacturers recommend certain dosage intervals for each drug and as near as possible these should be adhered to. Some drugs *must* be given at certain intervals to ensure adequate and continuous levels of the drug in the body, for example, phenobarbitone. Changes should only be made under the direction of the veterinary surgeon.

Right route

Unless given by the correct route of administration, the amount of drug that reaches the target tissues in the body can be dramatically reduced or altered. Once again, every drug manufacturer will recommend the suitable route for each drug. The general classification for drug administration is as follows:

Table 10.1 Routes for administering drugs.

Route	Site	Notes
Oral	By mouth	Convenient – can be administered by owner. Not suitable for vomiting animals
Intravenous route	Cephalic vein, lateral saphenous vein, jugular vein	Drug is introduced directly into the bloodstream. Fastest distribution of drug to its site of action. Highest peak plasma concentration than any other route However, drug concentration decreases more rapidly as absorption is more instant Preparations for intravenous route are solutions – drug is dissolved in water or other solvent. Always inject i/v solutions slowly Drugs which are irritant to tissues are preferably injected i/v
Intramuscular route	quadriceps, lumbodorsal, (triceps)	The hamstring and gluteals should be avoided due to close proximity of the sciatic nerve Large amounts of fluid may be painful if injected i/m (max. 5 ml in dogs, 2 ml in cats) Depending on the drug, effect will take place 20–30 minutes after injection The drug diffuses from site of injection by dissolving in the surrounding tissue fluid and is then absorbed into the local capillaries and lymphatic system When injecting, withdraw plunger on syringe to check the needle is not in a blood vessel
Subcutaneous route	Subcutaneous injections may be made into any areas of loose skin – the 'scruff' is the most common site	Least painful. Only non-irritant drugs should be used as local irritation may occur Absorption is much slower as there is a poor blood supply to the area. Depending on the drug, effect will take place 30–45 minutes after injection Large volumes of fluid may be injected s/c When injecting, withdraw plunger on syringe to check the needle is not in a blood vessel

- oral administration (per os) – drugs given by mouth;
- parenteral administration – drugs given by injection;
- topical administration – drugs applied to the surface of the body.

See Table 10.1.

Procedures

Correct procedure to remove drugs from multidose bottles should be followed.

(1) Check 'best before' date on bottle (discard if out of date).
(2) Check the contents of bottle for cloudiness or foreign particles (discard if present).
(3) Clean rubber cap with isopropyl alcohol (except insulin and vaccines).
(4) Insert a 19 gauge needle into cap to vent the bottle and prevent pressure differentials.
(5) Insert syringe needle and invert the vial.
(6) Withdraw the prescribed amount of drug.
(7) Remove syringe needle from bottle.
(8) Replace needle guard and tap the syringe to dislodge any air bubbles.
(9) Expel air from syringe.
(10) Remove 19 gauge needle from bottle and discard into sharps box.

Correct procedure to remove drugs from single-dose ampoules should be followed.

(1) Check 'best before' date on box of ampoules (discard if out of date).
(2) Check the solution for cloudiness or foreign particles (discard if present).
(3) Tap the neck of the ampoule gently to ensure that all of the solution is in the bottom of the ampoule.
(4) Cover the neck of the ampoule with a swab and snap it open.
(5) Inspect the solution for glass fragments (discard if present).
(6) Withdraw the required quantity of solution, tilting ampoule if necessary.
(7) Replace needle guard and tap syringe to dislodge air bubbles.
(8) Expel air.

Other important notes about injecting drugs should be observed.

- Always swab injection site with a cotton wool swab soaked in 70% isopropyl alcohol or antiseptic solution to remove surface debris from skin and hair (exceptions include insulin and vaccinations).
- Before injecting subcutaneous or intramuscular drugs, pull back on syringe plunger. If blood enters the syringe, remove the needle and select a new injection site. This is because the presence of blood in the syringe indicates that a blood vessel has been entered and some agents may cause severe reactions if injected intravenously.

Administration of cytotoxic drugs

- Wear protective clothing (two pairs of gloves, apron, goggles).
- Prepare all necessary equipment for aseptic administration of drugs.
- Check dosage.
- Always administer chemotherapy drugs via an intravenous catheter to avoid accidental perivascular injection.
- Inspect the infusion line and check the patency of the route with a large volume of 0.9% normal saline.
- Ensure the correct administration rate.
- Constantly supervise the patient and frequently check the vein for swelling and/or leakage at the site.

- Stop infusion if animal appears to experience pain at injection site.
- If the patient shows any evidence of interfering with the catheter or infusion line, apply an Elizabethan collar.
- Flush infusion lines and catheters with 0.9% saline between drugs and/or after administration.

Precautions:

- Put up warning signs on kennels and any equipment used. 'Toxic tape' is useful to label kennels, drip bags and clinical waste bags.
- Wash hands frequently.
- Wear disposable protective gloves and aprons when cleaning out kennel and handling the patient's excreta (wear disposable shoe covers if there is a danger of urine contaminating shoes).
- Wear protective clothing when handling the animal and cleaning out the kennel.
- Dispose of all clinical waste including kennel excreta and newspaper, infusion lines, syringes, swabs, etc. into 'high-risk' clinical waste sacks. Double bag all waste and dispose in the normal way for incineration.
- Double bag all bedding and label with 'cytotoxic contamination – wear gloves to handle'. Machine wash as normal.
- Take patient to a designated outdoor run to urinate. Due to the high rate of fluid administration during the therapy, patients will need to urinate more frequently.
- Rinse run with copious amounts of water after each use.
- Wear protective overshoes or specially designated Wellington boots.

DISPENSING DRUGS

The VN should be able to dispense drugs prescribed by the veterinary surgeon to clients, ensuring that all medication is correctly labelled and the dose is correctly calculated.

Points to remember when dispensing drugs

Name of the drug?

Drugs are generally referred to by three different names.

- Chemical name – used by chemists and pharmocologists, not generally used in veterinary practice, e.g. D-α-amino-p-hydroxybenzyl-penicillin trihydrate.
- Generic name (or non-proprietary name) – the more concise name given to the specific chemical compound, e.g. amoxicillin.
- Trade name (or proprietary name) – unique to each manufacturer for its particular brand of drug, e.g. Clamoxyl™.

In what form can the drug be dispensed?

It is useful to be aware of different formulations of the same drug. Liquid suspensions can be easier to administer than tablets. Knowledge of whether the drug can be given with food is essential and whether any food types might affect the absorption of the drug. See Table 10.2.

Table 10.2 Forms in which drugs may be dispensed.

Form of drug	Variations
Tablet – contains the drug in a powdered form, compressed into discs	*Sugar-coated* to make administration easier by hiding the bitter taste of the drug, and helps identification
	Capsules contain the drug in powdered or granular form within a hard gelatin outer case. The gelatin hides a bitter taste and is easier to swallow
	Enteric-coated tablets contain a special covering over the tablet to protect the drug from the harsh, acidic environment in the stomach and prevent it dissolving until it enters the intestine
Liquids	*Solutions* contain the dissolved drug in a liquid medium. This mixture does not settle out or precipitate if left to stand
	Suspensions contain the drug particles in the liquid, but they are suspended rather than dissolved. This means the drug settles at the bottom of the container if left. It is essential to shake the container to resuspend the drug thoroughly
	Emulsions contain a mixture of the drug and liquid fat or oil
	Gels contain the drug in a semi-solid mixture
	Syrups are solutions of the drug in water and sugar
Topical applications	*Liniments* contain the drug in an oil-based solution designed to be rubbed onto the skin
	Creams contain the drug in a semisolid form in oil or fat and water, designed to liquefy at body temperatures and spread easily
	Ointments contain the drug in a semisolid form in a greasy mixture, insoluble in water
	Pastes are as above, but are most often packaged in large plastic syringes for oral administration, e.g. cattle wormers
Injectable drugs	*Ampoules* contain the drug in a small, glass containers for single use
	Single dose vials contain enough of the drug for one single administration, e.g. vaccines
	Multidose vials are glass bottles of drugs with rubber stoppers. The drug is removed from the vial by inserting the needle through the rubber. The rubber stopper of multidose vials should be disinfected before the needle is inserted to prevent contamination of the contents of the bottle

Table 10.3 Drug containers.

Container	Medicine
Coloured glass fluted bottles	Medicine for external application only (shampoos etc.)
Plain glass bottles (clear or amber)	Oral liquid medicines
Wide mouth jars	Creams, dusting powders, granules
Plastic or glass tablet pots with childproof lid	All oral tablets and capsules
Cardboard cartons or wallets	'Blister-pack' tablets, sachets

Are there any contraindications?

Every drug comes with a *data sheet*, which contains a large amount of information about that particular drug. The data sheets include the following information.

- Indications: the approved uses for the drug.
- Dose that is recommended: the quantities and the frequency of administration.
- Adverse effects: the effects of the drug other than its intended beneficial effect.
- Drug interactions: highlighting other drugs that may interfere with the effects of the drug in question.
- Contraindications: conditions when the drug should not be used (e.g. during pregnancy).
- Extra-label or off-label uses: when the drug is used in species or for uses other than those intended by the drug's manufacturer.

What should the drug be put into?

The Council of the Royal Pharmaceutical Society recommends that certain containers be used for the different types of medicines. Paper envelopes and plastic bags are unacceptable. Suggested safe containers are listed in Table 10.3.

What should be written on the label?

The Medicines Act and the Medicine Labelling Regulations state the legal requirements for labelling dispensed drugs. All labels must be legible (preferably computer generated and typed) and indelible. The following details should be included.

- The owner's name and address.
- Date.
- Product name and strength.
- Total quantity of the product supplied.
- Instructions for dosage.
- Practice name and address.
- 'Keep out of reach of children'.
- 'For animal treatment only'.

Additional information may include:

Mr M Welsh MRCVS
Benmore Small Animal Clinic
Wickham Place

10th May 2010

For Ms Orpet's cat Ezekiel
Rose Cottage

10 Ampicillin 50mg
Give one tablet twice a day for five days

For Animal Treatment Only

Keep Out of Reach of Children

Fig. 10.1 Dispensing label.

- 'For external use only' on labels for products for topical use.
- 'WEAR GLOVES when handling drugs'.
- 'Give with food'.
- 'Give before food'.

An example is shown in Fig. 10.1.

Which dispensing and prescribing category is the drug in?

Retail supply of veterinary drugs is restricted to four categories:

- POM-V (Prescription Only Medicines – Veterinary). Controlled Drugs (CD) are a subcategory of POM-V's.
- NFA-VPS (Non-Food Animal – Vet, Pharmacist or SQP).
- POM-VPS (Vet, Pharmacist or SQP).
- AVM-GSL (Authorised Veterinary Medicine – General Sales List).

NOTE: Drug classifications are different in the Republic of Ireland.

Controlled Drugs

Controlled Drugs are regulated by the Misuse of Drugs Act 1971 and the Misuse of Drugs Regulations 1985. These regulations classify such drugs into five schedules (Table 10.4), numbered in decreasing order of severity of control.

What precautions should be taken?

Safe handling of drugs and knowledge of which ones may pose a health risk to the handler are essential. The regulations governing the safe handling of drugs are the Health and Safety at Work Act 1974 and the Control of Substances

Table 10.4 Controlled Drug schedule.

Schedule 1	Addictive drugs, e.g. cannabis, LSD, mescaline
Schedule 2	Opiate analgesics, e.g. morphine, etorphine, Immobilon, pethidine, fentanyl, amphetamine, cocaine, diamorphine (heroin)
Schedule 3	Barbiturates, e.g. pentobarbitone, phenobarbitone, opiate analgesics e.g. buprenorphine, pentazocine
Schedule 4	Benzodiazepines, e.g. diazepam
Schedule 5	Certain preparations of cocaine, codeine and morphine that only contain small quantities of the drug, e.g. codeine cough linctus, kaolin and morphine suspension

Hazardous to Health (COSHH) Regulations. Every veterinary practice must have produced a COSHH assessment sheet for *all* substances that staff may come into contact with. This document should:

- identify the potential risk to handlers;
- state the precautions handlers should take to avoid exposure;
- list the steps to take in the event of accidental exposure.

General rules for safe handling of drugs

The Health and Safety Regulations state that it is the responsibility of *each individual* to take care of their own health. This means that VNs should take safety into their own hands, not rely on being told by others in the workplace.

- Wear gloves when handling *all* drugs (many capsules and tablets can be safely handled without gloves, however it is good practice to wear gloves when handling all types of medicine).
- Use a triangular tablet counter when counting tablets to reduce contact with drugs.
- Wear face masks and eye goggles when handling powders and/or aerosol sprays.
- Dispose of used needles and syringes immediately after use. Dispose of contaminated packages and bottles appropriately.
- Keep all food and drink away from the pharmacy and wash hands before eating, drinking or smoking.
- Thoroughly clean surfaces after working with drugs in that area.

LEGAL ASPECTS OF MEDICINES AND PRESCRIBING

The following regulations and acts are concerned with how veterinary practices buy, sell, store, prescribe drugs and the associated safety issues:

- Veterinary Medicine Regulations;
- Misuse of Drugs Act 1971 (see section above on Controlled Drugs);

- Misuse of Drugs Regulations 1985 (see section above on Controlled Drugs);
- Health and Safety at Work Act 1974;
- Control of Substances Hazardous to Health Regulations 2002.

REFERENCES AND FURTHER READING

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SECTION 3

MANAGING THE OPERATING THEATRE

Chapter 11

An Introduction to Theatre Management

THE ROLE OF THE VETERINARY NURSE IN THEATRE

Operating department nursing was described as the first (human) nursing specialisation. Towards the end of the 19th century, this was a very prestigious role where the nurses' responsibilities included creating the ideal environment, preventing infection and providing appropriate equipment and providing care for the patient during surgery.

The operating room can provide VNs with a varied and challenging range of professional activities. Aside from the actual management and preparation of the surgical environment and equipment, the VN's role can extend to include a range of patient care and treatment activities. Well organized and efficient veterinary practices can benefit from making full use of the abilities of well trained and skilled nurses; ranging from the preparation of the patient and team for surgery, skilled assistance during surgery and performing minor surgical procedures.

Many of the larger clinics and teaching hospitals employ a VN to work specifically in this area because they identify that theatre efficiency can be maximised by ensuring that a highly qualified and specialised person is involved with the day-to-day running of the area. In terms of general practice, the VN can use his or her expertise to raise standards of hygiene and sanitation in the theatre and organise the area and other veterinary colleagues to maximise operating room order and efficiency.

Responsibilities include:

- maintaining theatre hygiene;
- cleaning surgical equipment;
- sterilising equipment;
- maintaining surgical instruments;
- assisting with surgical list planning;
- surgical preparation of the patient;
- intraoperative patient care;
- assisting during surgical procedures;
- performing minor surgical procedures (according to amendment to the Veterinary Surgeons Act in 1991).

THE OPERATING ROOM AND ENVIRONMENT

In an ideal situation, the operating room and environment should be spacious, purpose built and used specifically for the purpose of operating and nothing else. It should contain minimal equipment so cleaning can be thorough. It is an area where patients can be vulnerable to infection so logging of cases can enable potential hospital-acquired infections to be acted upon immediately.

Methods of preventing infection in theatre

- Routinely disinfect theatres.
- Restrict movement through the theatre area.
- No through traffic.
- Surgeons to scrub and glove effectively.
- Check sterilisation methods regularly.
- Everyone in theatre to wear clean scrub suits and theatre shoes.
- Use appropriate order for surgical procedures, i.e. clean operations before dirty operations (e.g. orthopaedic operations before dental operations, bitch spay before gastrointestinal surgery).
- Ensure adequate clipped area on the patient.
- Aseptic operative technique.
- Keep surgical time as short as possible.

Chapter 12

Preparation of the Theatre for Surgery

Maintenance of asepsis in theatre is very important, therefore cleaning protocols should be adhered to.

PROCEDURE

Every day before the surgical session:

- damp-dust the operating room with disinfectant (all surfaces and equipment);
- check the theatre list and set up equipment ready for use;
- collect all instruments that may be required for the operation;
- ensure the anaesthetic machine and circuits are working.

Between surgical cases:

- remove and clean instruments and equipment;
- remove all waste materials (tissues, swabs, empty packets);
- clean and disinfect 'kick-about' bowl;
- clean and disinfect operating table and instrument trolley;
- clean gross dirt (blood, etc.) from floor;
- set up for next operation.

After the surgical session:

- remove all instruments and equipment for cleaning and sterilising;
- clean and disinfect all surfaces, lights, equipment and walls;
- disinfect an area of the floor so that the equipment may be rolled on to this spot whilst the rest of the floor is cleaned;
- restock for next session.

It is useful to obtain swabs for bacteriology from the operating room environment periodically.

CLEANING EQUIPMENT AND SUPPLIES

Wet-vacuums (with disinfectant) are preferable to mops when washing floors but not always practicable. Mops are a potential source of infection. It is

important to colour code the mops that are used in the practice, ensuring the same mop and bucket is kept for theatre. They must be rinsed thoroughly after each use, and then soaked in disinfectant for 30 minutes or hot wash laundered before standing on end to dry. Mop buckets should also be cleaned and stored upside down so they can dry properly. Disposable gloves should be worn when cleaning theatre and equipment as they help to prevent infection and protect against irritant/harmful effects of some cleaning and disinfectant products. Cloths and sponges should preferably be disposable to minimise contamination, otherwise launder and dry them daily.

Chapter 13

Sterilising Equipment for Theatre

Sterilisation is the destruction of all microorganisms and spores. Surgical instruments and other equipment used during surgery must be sterile before use. There are various methods that can be employed to ensure effective destruction of pathogenic microorganisms and their spores and it is the responsibility of the VN to be familiar with the various techniques and their suitability for different surgical equipment.

STERILISING METHODS

The method by which items may be sterilised will depend on:

- the material, e.g. plastic, fabric, metal;
- the quantity and size of items;
- financial constraints and sterilising equipment available.

Sterilising methods may be divided into six main categories:

- dry heat: hot air oven;
- moist heat: autoclave;
- gaseous: ethylene oxide;
- chemical: gluteraldehyde;
- irradiation: gamma radiation;
- gas plasma: hydrogen peroxide.

Dry heat

Very high temperatures kill microorganisms and their spores by destroying cell protoplasm. This method of sterilisation has limited use, mainly because the high temperatures that are required cause damage to many materials. It is, however, the method of choice for fine surgical equipment where autoclaving may blunt the fine points. It can also be useful for sterilising glassware (in the laboratory) and waxes and oils, which cannot be sterilised using moist heat. The oven requires time to heat up and cool down in addition to the sterilising time. The items are placed (unpacked) on to metal trays within the oven and left for a minimum of 60 minutes at 180°C (Table 13.1). The oven is heated by an electrical element and should preferably be fan-assisted to prevent cold spots within the oven leaving some items unsterilised.

Table 13.1 Times and temperatures for sterilising items.

Temperature	Time at this temperature
160°C	120 minutes
180°C	60 minutes

Table 13.2 Autoclave temperature, time and pressure combinations.

Temperature	Time	Pressure
121°C	15 minutes	15 psi (103 kPa)
126°C	10 minutes	20 psi (138 kPa)
134°C	3.5 minutes	30 psi (207 kPa)

Moist heat

Microorganisms are destroyed by wet or dry heat. Bacterial spores show a greater resistance to dry heat than to moist heat. The presence of moisture coagulates the critical cellular proteins and destroys them at lower temperatures. When water boils at 100°C it turns into steam. By raising the pressure within a chamber, the temperature of steam is raised. The higher the pressure, the higher the temperature and so the length of time required for sterilisation is reduced (Table 13.2).

A steam jacket surrounds the central sterilising chamber of an autoclave. During the sterilising process, the pressure in the jacket is raised before steam is introduced into the main chamber. Pre-existing air in the chamber is pushed downward by the steam and expelled from the system through vents to the outside. When all the air is removed and the desired pressure reached, the heat produced by the steam penetrates the innermost layers of the equipment packs.

Gaseous

Ethylene oxide liquid is contained in a glass ampoule which, when opened, releases a gas that kills microorganisms by a process called alkylation (essential cellular metabolic reactions are blocked). It is particularly useful for equipment that is damaged by moist or dry heat. Items are individually packaged and placed into a polythene liner bag that is placed into the special Anprolene chamber. The glass ampoule is positioned in the middle of the bag and the top is broken off to release the gas. The door of the chamber is then closed and locked for the required period of time (12 hours' sterilisa-

tion time plus 2 hours' purge cycle). Strict safety regulations control the use of Anprolene, and the manufacturer's safety recommendations and COSHH Regulations must be observed.

Chemical

Gluteraldehyde liquid is capable of destroying microorganisms and their spores. Items are placed into a container filled with a gluteraldehyde solution and submerged for up to 24 hours, depending on the manufacturer's recommendations. It is essential to rinse the equipment thoroughly afterwards with sterile saline because of the toxic and irritant effect of gluteraldehyde. Gloves, face masks and protective eyewear should be worn whilst handling. This method is suitable for materials that will tolerate little or no heat, e.g. certain plastic tubes and fibreoptic equipment.

Irradiation

Beta and gamma rays may be used to sterilise most materials, with the exception of certain plastic and rubber combinations. The equipment required to generate the radioactive rays is very expensive and there are strict safety controls that restrict its use exclusively to industry. Many of the prepacked surgical materials such as suture materials and gloves are sterilised by this method.

Gas plasma

When an electric or magnetic field is applied to a solution (hydrogen peroxide) a partially ionised gas forms. This comprises of ions, electrons, ultraviolet photons and reactive neutrals. The plasma gas causes destruction of lipids, DNA and other cell components by producing hydroxyl free radicals. The process takes about an hour but has the advantage that a range of items normally destroyed by heat can be sterilised. It is not suitable for cloth or paper however.

STERILISING INDICATOR SYSTEMS

Successful sterilisation will depend on various factors such as the correct preparation of the packs, correct loading and operation of the steriliser and adequate times for sterilisation. It is important to use an indicator with every load to ensure that sterilisation of the equipment has actually occurred. Available methods are shown in Table 13.3.

All equipment and instruments should be cleaned, dried and lubricated as necessary prior to packaging in the most appropriate wrapping material. The ideal wrapping material should be permeable to steam or gas but not to microbes and it should be flexible and resistant to damage. Different types of wrapping material are shown in Table 13.4.

Table 13.3 Sterilising indicators.

Method	Action
Bowie-Dick tape	Bowie-Dick tape is used to seal drape and instrument packs for autoclave sterilisation. It is impregnated with chemical stripes that change to dark brown when an adequate temperature has been reached (121°C). The tape responds only to temperature changes, irrespective of times and pressures, which makes this method an unsuitable way of determining if sterilisation has been achieved. Additional methods should be used at the same time
Browne's tubes	Browne's tubes are small glass tubes partially filled with a liquid that changes colour from orange to green in response to the correct temperature being maintained for a required length of time. Browne's tubes are available for use in both autoclaves and hot air ovens and different tubes respond to the various ranges of temperatures (121, 126, 134 and 180°C). The appropriate tube should be selected for the sterilisation method being used
TST strips	Time Steam Temperature (TST) strips are strips of card with an indicator spot that changes colour (yellow to dark blue) in response to time, temperature and pressure. They are suitable for assessing effectiveness of sterilisation in the autoclave. It is important to use the correct strip for a particular time/pressure/temperature setting. Place in the centre of the pack
Indicator spots	Commercially available plastic/paper pouches are available for packing items for sterilisation by autoclave and ethylene oxide. Each bag is printed with two indicator spots, one is sensitive to ethylene oxide and the other is sensitive to moist heat. They change colour when exposed to the sterilisation process: Autoclave spot changes from pink to brown Ethylene oxide spot changes from blue to yellow
Spore tests	These are strips of paper that have been impregnated with dried bacterial spores. Following the autoclave cycle, the spore tests are sent for culture. If sterilisation has been effective, there will be no growth of any microorganisms. It is important to use additional methods of assessment at the same time because results from this test are not immediate

The choice of packaging depends on:

- the size of the steriliser;
- cost;
- size of items to be packed.

Method of packaging

Instruments should always be placed in the packaging so that when the package is opened, the handles are facing out first. This enables the surgeon to take the instruments by the handles. Instruments should be placed into the autoclave bag so that the handles are facing the opening tabs. Instruments and packed equipment should not be 'dropped' on to the instrument trolley

Table 13.4 Types of wrapping materials.

Nylon film	Nylon film is designed particularly for use in autoclaves, it comes on a roll, in a variety of widths that can be cut to the desired length. It is relatively cheap and reusable although it starts to become brittle after several uses. It is transparent, which allows easy identification of the instruments inside, and is waterproof
Peelable pouches (seal and peel) (plastic film and paper)	These are disposable pouches which are naturally more expensive than film. However, they have many benefits, including: • they are easy to open; • they come in a variety of sizes; • they are transparent (on one side); • they can be used with autoclave or ethylene oxide; • they have sterilising indicator spots on the packet.
Paper	Large sheets or rolls of a slightly elastic, crepe paper are available for wrapping around kits of instruments. The paper is water resistant and can be cut to the required size
Textiles	Certain cotton textiles (such as drapes and towels) can be used to wrap instrument packs. The advantage is that they are reusable and conforming. However, cotton is not water resistant and it is better to use cotton with a paper outer covering
Metal drums	Metal drums of various sizes are available and equipment may be placed directly inside these and sterilised. They are expensive to buy initially, however they can be reused forever. Their main disadvantages are that they are too large for most practice table-top sterilisers and they are often multi-used, therefore the rest of the kit is contaminated when they are opened
Boxes and cartons	Corrugated plastic boxes with lids are available in a range of sizes for use in autoclaves. They are water resistant, reusable for many years and are particularly useful for complete kits and those containing specialised collections of equipment (e.g. orthopaedic kits)

but should be opened and passed to the surgeon or scrubbed assistant. Sharp tips or ends of equipment should be covered with a protective cover. Special autoclave rubber tips are available or a gauze swab can be wrapped over the end. Plastic syringe cases can be autoclaved and they make useful containers for needles etc.

The method of folding a gown for sterilisation.

(1) Lay the gown on a flat surface, with the outside of the gown facing upwards.
(2) Smooth the front ties and place down the gown.
(3) Fold the sleeves into the centre of the gown.
(4) Fold one back flap into the centre of the gown.
(5) Fold the second back flap into the centre of the gown.
(6) Smooth down the back flap ties.
(7) Fold into an accordion pleat.
(8) Place folded gown into sterilising package.

Labelling of packs

All packages must be labelled with:

- date;
- name of piece of equipment or name of kit;
- name or initials of the person who packed the equipment.

Storage of packs

The way equipment is packed and stored will affect the shelf-life of packs. A separate area should be available for storage of sterilised equipment, which is dust free and away from contaminated articles, preferably in closed cabinets. There are varying thoughts on how long the packs stay sterile but they can usually be considered sterile for up to 1 year, after which time they should be re-sterilised.

Extremes of temperature, humidity and excessive handling will shorten the shelf life of the packs. The most recently sterilised items should be placed behind older packs so that the older packs can be used first.

Chapter 14

Care of Surgical Equipment

CARE OF SURGICAL INSTRUMENTS

The role of the VN is extremely important in the care and maintenance of instruments and theatre equipment. If cared for, maintained properly and used only for their intended purposes instruments can last for up to 10 years.

Most instruments are made of stainless steel, which is an alloy of several metals: iron, carbon and chromium. The chromium element is what makes the steel resistant to corrosion. In their manufacture, instruments go through various processes to increase their durability:

- the degree of hardness is achieved by subjecting the alloy to various degrees of heat;
- they are placed into nitric acid which clears the metal of foreign matter or debris and also helps to form a protective corrosive-resistant chromium oxide layer.

There are three different finishes for instruments:

- a mirror finish, which has been highly polished and makes the instrument very resistant to corrosion;
- a satin finish, which is designed to avoid the glare of the mirror finish instruments;
- an ebony finish, which eliminates glare completely.

Tungsten carbide may be inserted into the tips and jaws of some instruments to provide an extremely hard-wearing surface, which can be replaced. These instruments usually have gold handles. Blue-coloured instruments, made of titanium, are used for some fine ophthalmic instruments because they are very light and hard wearing.

Colour coding instruments with special autoclavable tape will ensure that instruments are packed into their appropriate kits, and reference cards listing specific instruments for each surgical procedure will make sure that all instruments are gathered up prior to surgery.

Care and maintenance of instruments:

- Instruments must only be used for the purpose for which they were intended.
- Handle instruments carefully, never throw or drop them.
- All new instruments must be cleaned, lubricated and sterilised before use.
- Check hinged instruments for movability. All box joints should work smoothly.
- Send blunt cutting instruments and drill bits away for re-sharpening.
- Place protective tips over sharp and delicate instruments when they are not in use.

Procedure for cleaning instruments

(1) Never leave instruments dirty for long periods of time. Blood causes corrosion and staining of the instruments.
(2) Place instruments in cold or lukewarm water with a mild detergent (e.g. washing-up liquid) or preferably into a special surgical cleaning solution such as Medigene™, as soon as possible.
(3) Never leave the instruments to soak for long periods of time. Tap water may leave deposits on the instruments and sterile saline corrodes stainless steel. Ideally use distilled and deionised water or surgical cleaning solution.
(4) Wearing gloves, use a soft bristle brush to remove gross debris, pay particular attention to the ratchet mechanism, and the jaws of forceps.
(5) An ultrasonic cleaner may then be used if available (up to 90% of debris is removed after a 5 minute cycle). Always place the instruments with their joints open into the cleaner.
(6) Rinse instruments in cold water to remove detergent agent.
(7) Dry the instruments as much as possible, paying attention to joints and ratchet mechanisms. Place in warm area if possible to help dry (on top of boiler or heated cabinet if available).
(8) Lubricate instruments especially joints and ratchets with instrument milk or spray. If instrument milk is used, the instruments do not need to be dried first.
(9) Check the instruments thoroughly before sterilising, looking for any damage to joints, blades or ratchet mechanisms.
(10) Instruments should not be marked by etching methods as this will destroy the protective chromium layer.

COMMONLY USED INSTRUMENTS

Nurses should be able to recognise commonly used instruments and understand their use. A comprehensive list is given in Table 14.1.

CARE OF OTHER SURGICAL EQUIPMENT

There are pieces of surgical equipment which help the surgeon by saving time and making life far easier. If used and maintained correctly, the long-term benefits outweigh the cost disadvantages.

Table 14.1 Common instruments.

Instrument type	Example	Use
Scalpel handles	Bard-Parker	For use with size 10, 11, 15 and 20 blade sizes
Needle holders	Mayo-Hegar Olson- Hegar Gillies Mcphail Bruce-Clarke	Should be able to hold on to an appropriately sized needle without allowing rotation. Combination of holders and scissors (Gillies, Olsen-Hegar)
Scissors	Metzenbaum Mayo (straight or curved) Lister (bandage) Spencer (stitch)	Use Mayo for dense tissue and Metzenbaum for more delicate dissecting Specialist types (Potts) angled for cardiovascular work Classified by their blade points: sharp/blunt sharp/sharp blunt/blunt
Haemostats	Halstead Mosquito Kelly Crile Spencer-Wells	Range from fine mosquito to sturdier forceps often used for bone manipulation Always check alignment and ratchet mechanism
Tissue forceps	Allis Babcock Lane Doyen bowel clamp	Allis are fairly traumatic, do not use on skin or hollow organs. Babcock are slightly less traumatic
Thumb forceps	Adson	
Dressing/tissue forceps	Semkin Brown-Adson DeBakey Cooley Jeans	Plain or with teeth to grip the tissue. DeBakey and Cooley are atraumatic for soft tissues and cardiovascular work
Towel clips	Backhaus Jones (similar to cross action) Cross action	Secures drape to skin (should be sharp). Do not remove once it has been placed as it breaks sterility
Pin vice	Jacobs chuck	Holds Steinman pins and Kirschner wires
Bone saw	Gigli saw	
Retractors: hand-held	Hohmann Langenbeck Malleable	Allow adequate visualisation in a body cavity or joint. Care must be taken not to trap intestines or nerves. Swabs may be used with abdominal retractors
Retractors: self-retaining	Gelpis Wests Travers Gosset Finochietto Turvier Balfour	

Table 14.1 *Continued*

Instrument type	Example	Use
Bone-holding forceps	Lowman Spin (speed) lock Reduction	Used to manipulate and hold fragments of bone in place. With or without ratchets
Rongeurs	Lempert Stille Luer Kerrison	Bone 'nibblers'. Must be used correctly or the jaws will blunt and become pitted Kerrison rongeurs are used for fine spinal work
Bone-cutting forceps	Liston Ruskin	Used for reflecting the muscle from the bone
Periosteal elevators	ASIF Freer	
Osteotomes	Lambotte	Both used for cutting bone. Used with a mallet
Chisels	Stille	
Curettes	Volkmann Sprat	Designed for scraping tissue from cavities or collecting cancellous bone grafts. Fine curettes are used for removing calcified vertebral disc material
Trephines	Michel	Used for bone biopsies or for collecting cancellous bone grafts
Ovariohysterectomy hook	Snook Covault	
Suction piece	Yankauer Frazier Ferguson Poole	For removing fluid or blood from the surgical field. Some come with an inner stylet to prevent blockage

Electrocautery (diathermy)

Haemostasis during surgery can be achieved in various ways, including application of gauze swabs, haemostat forceps and vascular cauterisation.

Vascular cauterisation may be performed using an electrocautery (diathermy) unit. Bleeding vessels (arteries up to 1 mm and veins up to 2 mm) are sealed using a controlled electrical current, which coagulates and seals the bleeding end of the vessel.

Advantages include:

- reduction in surgical time;
- reduction of overall blood loss;
- increased visualisation of the surgical field.

Different manufacturers produce various types of unit, ranging from simple battery-powered units to extremely powerful and sophisticated electrocautery models. The patient must be properly grounded during use otherwise the electricity used for the cauterisation will travel along the path of least resistance and burning or electrical shock to the patient or surgeon can

occur. Alcohol and other highly flammable materials should not be used when electrocautery is carried out. Deposits of blood and tissue can build up on the tips of the probes and it is important that they are cleaned regularly throughout the surgery with a swab to ensure good contact is maintained. Some electrosurgical units may be switched to a 'cut' setting. Some surgeons prefer this method of cutting skin because it is faster and less bleeding occurs, however, there is evidence to suggest that incisions made this way take longer to heal. Small warts and tumours may also be removed this way using a circular probe end to scoop out the tumour from the surrounding tissues.

Probes and leads can be unplugged from the main unit and sterilised using the manufacturer's instructions, usually by steam or ethylene oxide.

Cryosurgery

Mammalian cells are destroyed when they are frozen to a temperature of –20°C (–4°F). Cryosurgery is a method of destroying unwanted tissue by the application of very cold temperatures to the desired area. It is particularly useful for treatment of conditions such as:

- squamous cell carcinoma;
- trichiasis and distichiasis;
- lick granuloma, anal furunculosis;
- perianal fistulas.

There are two cryogens widely available:

- liquid nitrogen;
- nitrous oxide.

Liquid nitrogen is a liquid and is the most versatile cryogen available. It can be used in a spray gun system or using a probe. Some people dip cotton-bud like applicators directly into the liquid nitrogen and then apply to the tissues. The liquid nitrogen can be delivered directly to the practice and stored in vacuum-insulated containers until it is ready to be used. Some of the liquid can then be poured into a cryosurgical spray gun when required. Care must be taken when handling liquid nitrogen; if accidentally splashed onto the skin, it freezes areas of contact. Protective goggles and gloves should be worn when pouring it into the spray gun.

Nitrous oxide is a gas and is supplied in blue gas cylinders (often used as an analgesic agent during gaseous anaesthesia). The gas is released under high pressure through a small orifice inside a metal probe via a reducing valve and tube system. This action produces probe temperatures of –89°C (–128.2°F). This frozen probe is then applied to the tissues. Probe freezing is generally easier to control than spray but is less lethal to tissues and can therefore only be used for small cutaneous lesions (<3 cm diameter). Very little patient preparation is required. The animal should be anaesthetised and the area immediately surrounding the target tissue should be clipped to allow easier visual inspection of the tissues during the freezing process. Gentle cleansing of the skin to remove debris and dirt may then be performed.

Nitrous oxide has health and safety implications and steps should be taken to prevent environmental contamination.

General technique

The tissues to be destroyed should be frozen and thawed at least twice during a cryosurgical session. Some surgeons apply three freeze–thaw cycles. If a probe is used it is applied directly to the tissues. A spray is held at a distance of 2.5–5 cm from the lesion. During the application, a creamy yellow colour 'frostball' is created in the affected tissues, and the size of this should be closely monitored to ensure that all of the affected tissues have been frozen. The frostball should be approximately 1 mm or larger than the lesion and should be maintained for a period of 30 seconds only. When the first frostball has thawed, the second freeze can be applied to the tissues.

Postoperative care

The postoperative changes that occur in the cryosurgical patient are quite profound and can distress an unprepared owner. It is essential that the VN is aware of the postoperative changes and advises the owner at the time of discharge.

- Swelling. Occurs within hours of treatment but should resolve within 48 hours.
- Haemorrhage. Some ulcerated type lesions will haemorrhage following cryosurgery. Temporary dressings may be applied if this is the case.
- Necrosis. The whole point of cryosurgery is to destroy tissues by freezing. This of course results in tissue necrosis and sloughing which is most often the most disturbing aspect of cryosurgery visually. The tissue usually sloughs off in about 10 days to reveal a granulating wound.
- Depigmentation. Freezing destroys melanocytes and hair follicles so the new tissue is a different colour for some time afterwards. The area always remains hairless.
- Odour. If freezing involved mucous membranes or if the patient interferes with the site, the scab becomes moist and may smell offensive. If this is the case, the area can be cleaned daily with a mild soapy water solution.
- Self-trauma. The area must be protected from self-trauma by the use of Elizabethan collars or other suitable method.

Care of cryosurgical equipment

The probes may be removed and washed in lukewarm water. They may then be sterilised as per the manufacturer's instructions. Any excess liquid nitrogen should be returned to the storage container (taking care to wear the correct personal protective equipment). The unit can be wiped over with dilute disinfectant solution.

Surgical lasers

LASER stands for Light Amplification by the Stimulated Emission of Radiation. Understanding how laser works is highly complicated, but in short

the device transfers light of various frequencies into an extremely intense, small and nearly non-divergent beam of monochromatic radiation. It is capable of mobilising immense heat and power when focused at close range. Laser is primarily used as a tool in surgery but is also used as a modern version of acupuncture where it provides a quick, painless and non-invasive method of point stimulation. The main use of laser in veterinary situations is when extremely precise incisions are required and for malignant tumour removal. The main considerations when using laser therapy are the safety aspects concerning its use. Water buckets and CO_2 fire extinguishers must be easily available during the procedure. A warning sign with the laser logo should be posted outside the operating room door as a safety warning. Ocular damage to the patient and operating team must be avoided by wearing specialised full-coverage goggles. Special blackened instruments should be used to prevent reflection of the laser beam from instruments on to skin causing burns. Laser machines should be operated in low ambient temperature, with low humidity and dust-free environment for longer life of machines.

Endoscopy

An endoscope is an instrument used for direct visual inspection of hollow organs or body cavities. There are two main types of endoscope: flexible and rigid. Flexible endoscopes consist of a flexible fibreoptic tube, which contains glass-fibre bundles that are capable of transmitting light and images along the tube. This is attached to an eyepiece, with control knobs, air/water suction valves and biopsy channels. Some flexible endoscopes can be attached to a computerised video processor and television monitor, which enables multiple people to view the image and allows the images to be saved on to DVDs, CDs or memory sticks. Rigid endoscopes are much simpler systems consisting of a metal rigid tube containing a series of lenses or glass rods. The images bounce from the lens to the camera.

The VN's role in endosocopy is an important one, being fully competent in setting up the equipment and preparing the patient (e.g. prior to colonoscopy, administer enema), and help with obtaining biopsy samples if necessary during the scoping session. Correct care and maintenance of the equipment is essential. The fibreoptic bundles in the flexible endoscopes are extremely fragile and the VN must use extreme caution when handling these scopes. Broken fibres cause black spots on the image and are costly to repair. Scopes should be cleaned inside and out, first to remove any organic matter and secretions, and then sterilised using an appropriate method as recommended by the manufacturer. Suitable methods usually include cold sterilisation using gluteraldehyde solution or ethylene oxide. The equipment should always be returned to its protective storage or carrying case when not in use.

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SECTION 4

THE OPERATING ROOM – SURGICAL NURSING ACTIVITIES

INTRODUCTION

Any surgical procedure, however minor, carries a risk to the patient. For the VN this is an area where some of the most potentially harmful procedures are performed. It can also be where some of the most legally contentious activities occur, so it is essential that the nurse is not only fully trained and competent in these skills, but is fully aware of his or her legal and professional status and limitations. In particular the VN must be fully familiar with the RCVS Guide to Professional Conduct and the legislation relating to Schedule 3 of the Veterinary Surgeons Act 1966 (refer to Section 1). Schedule 3 of the Veterinary Surgeons Act 1966 specifically applies to the listed or registered VN's (or student nurse's) status with regard to minor surgical procedures. It stipulates that qualified, listed nurses can perform minor surgery (not involving entry into a body cavity) under the direction of the veterinary surgeon that is providing care for a companion animal. There is not a prescriptive list of procedures and therefore VNs must be aware of what constitutes an act of veterinary surgery and what may come under the Schedule 3 amendment. Appropriate training should take place and the nurse should be competent in carrying out the procedure.

This section deals with some of the procedures that a veterinary nurse will be expected to perform competently.

Chapter 15

Preparation of the Patient for Surgery

Preparation of any patient undergoing a surgical procedure, however minor or major, begins as soon as the animal arrives at the hospital. The VN may be responsible for many of the tasks carried out during the patient's stay at the hospital and it is essential that he or she has a thorough knowledge of the techniques employed and the pertinence of particular procedures to each surgical case.

ADMISSION TO THE HOSPITAL

An admissions checklist (this can be incorporated into the consent form) can be used to verify the patient's identity and proposed surgical procedure, to confirm that food and water have been withheld for the required length of time and have space for the owner's signature for consent to treatment and surgery. An estimate of the total cost of treatment can also be included. In addition further information regarding the animal's normal activities including feeding may also be obtained from the owner (see Chapter 2).

A general clinical examination and weight check should be performed and the results recorded. Any abnormalities should be reported to the veterinary surgeon. The patient may be exercised prior to admittance to allow urination and defecation. A note should be made of previous and current medication treatments (some medications may affect anaesthetic protocols or medication regimes). It may be useful in some cases to determine if the animal has had a blood transfusion in the past, as subsequent transfusion will require cross-matching.

General preparation

- Bath (day before).
- Food withheld for 12 hours (adult cats and dogs).
- Withhold water 2–3 hours prior to surgery (unless suffering from renal disease).
- Allow urination/defecation.
- Enema – if required.
- Placement of intravenous catheter – facilitates smoother induction, reduces risk of accidental perivascular injection, allows intravenous fluid administration and ensures a route for drugs in the event of an anaesthetic emergency.

PREMEDICATION

Pre-anaesthetic medication is frequently administered prior to induction of general anaesthesia for a number of reasons:

- to calm and control the patient;
- to provide analgesia throughout surgery;
- to reduce the amount of anaesthetic induction and maintenance agents required;
- to produce a smooth induction of anaesthetic;
- to reduce some of the unwanted side-effects of some anaesthetic agents;
- to ensure a smoother recovery from anaesthesia.

Many practices have standard premedication protocols and the VN may be given the task of calculating dose rates, drawing up drugs and administering the drug combinations. It cannot be emphasised enough that accuracy in all aspects of premedication administration is essential (see Chapter 10). Many of the drugs administered are classified as Controlled Drugs and VNs must be aware of the legal requirements for storage and record keeping for these drugs.

HAIR REMOVAL

Removal of fur should be carried out in a separate preparation room to the operating theatre to minimize contamination. Usually removal of fur is carried out whilst the patient is anaesthetised immediately prior to surgery. The noise of the clippers and the feel of the clippers against the patient's body is likely to disturb many animals. However, in some instances it may be desirable to ensure as short anaesthetic time as possible and clip long hair prior to anaesthetising the patient.

- Usually a number 40 Oster clipper blade is used.
- For delicate areas and around the eye, it is useful to use small clippers with a narrower blade. The use of a depilatory or razor is not recommended for use in animals because of their irritant effect on the skin.
- As a general guide, the clipped area should radiate at least 15 cm around the surgical incision (obviously depends on size of animal).
- The blade of the clippers should be kept flat against the skin to prevent excessive trauma to the skin.
- Blades should be sprayed frequently with a commercial blade coolant and lubricant to help prevent excoriation or clipper burns.
- The blades must be sharp and free of broken teeth to prevent skin abrasions.
- A sterile water soluble gel can be applied to any pre-existing open wounds or on to the surface of the eye as protection (unless intraocular surgery is being performed) and the fur clipped away from the wound. Fine loose hairs can then be washed away more easily during the cleaning stage.
- Loose fur can be removed from the skin, body and surrounding area by vacuuming or by using sticky tape around delicate areas.

PATIENT POSITIONING

- The veterinary surgeon will often decide how he or she wants the patient positioned. However, for many procedures there will be a standard position and the VN should be familiar with these.
- Positioning should only be carried out after the anaesthetic has been stabilised.
- Take care not to compromise circulatory or respiratory function. Obese animals or heavily pregnant animals are often positioned slightly to one side when positioned in dorsal recumbency.
- Placing the animal on a thermostatically controlled hot water bed or electric blanket will help to prevent hypothermia during surgery. Care must be taken with some heated pads, which may cause severe burning. Some operating tables are heated but it is essential that the recumbent animal does not suffer burns.
- When performing surgery on a limb, cover the distal part of the leg with a conforming bandage and suspend the leg from a drip stand. This will aid in the cleaning procedure and give the surgeon full access to drape the limb.

PREPARATION OF PATIENT'S SKIN FOR SURGERY

The VN must be aware that effective and careful disinfection of the skin surrounding the surgical site is a crucial step in helping to reduce the risk of postoperative infection. Whilst it is relatively easy to remove and kill the transient bacteria on the skin's surface with antiseptic solutions, it is more difficult to remove the resident bacteria. In normal circumstances, these subsurface bacteria do not cause a problem, however, during surgery (or any other invasive procedure) they can come to the surface and provide a source of infection. Thus the overall aim of skin disinfection is to eliminate transient skin flora and to reduce the resident skin flora without traumatising the skin. This can be achieved by applying suitable solutions, which will immediately remove and kill transient bacteria and provide some residual activity, which will help kill resident bacteria.

Skin preparation agents

There is a variety of solutions available specifically for use on skin; when deciding on a suitable protocol for preparation of the skin for surgery, it is important to select agents which have the following properties:

- a detergent to remove gross dirt and grease;
- ability to remain active in the presence of organic matter;
- an antimicrobial agent effective against a wide range of microorganisms;
- long residual effect;
- causes no or minimal skin irritation;

Commonly, chlorhexidine or povidone–iodine solutions are used for veterinary purposes. There have been many studies carried out to evaluate the efficacy of both agents, but there is still no conclusive evidence that indicates one is better in every way than the other. It is important, however, that each is used according to the manufacturer's specifications and recommendations.

Chlorhexidine is active against a wide range of bacteria and many fungi. It does not become deactivated when exposed to organic matter or alcohol and it has been shown to produce a 99% bacterial kill rate 30 seconds after application. Chlorhexidine has longer residual activity than povidone–iodine. Chlorhexidine is irritant to the cornea and should not be used around the eyes; instead, a 2% solution of povidone–iodine may be used followed by a sterile saline flush.

Povidone–iodine continues its bactericidal activity by the release of free iodine as it dries and the colour fades. As with chlorhexidine, it is effective against a wide range of bacteria and fungi. Its effectiveness is reduced by the presence of organic material (e.g. blood and fat) and is therefore best left as a final spray or paint solution after the initial preparation with chlorhexidine and isopropyl alcohol.

Isopropyl alcohol is effective against a wide range of Gram-negative organisms and some fungal spores. It is useful to apply isopropyl alcohol after scrubbing the skin because it removes the lather produced by the scrub solution and has a drying effect on the skin. Isopropyl alcohol should not be used on mucous membranes or on open wounds because it causes tissue necrosis.

Care must be taken to avoid wetting the patient too much during the preparation stage to reduce the risk of hypothermia. The evaporation effect of applying isopropyl alcohol also creates a cooling effect and it is probably better to avoid using it on very small or neonatal animals.

Antiseptic skin preparation protocol

Antiseptic preparation of the skin usually takes place after the patient has been positioned for surgery. However, it is advisable to carry out an initial skin scrub to remove excess hair and gross dirt before moving the patient into the operating room.

Initially in the preparation room the following procedures will remove gross dirt and debris from the site.

(1) Gather equipment: lint-free gauze swabs, bowl of warm water, antiseptic agent in pump dispenser, disposable gloves.
(2) Wear disposable gloves (reduces contamination of the site by the hands whilst at the same time protecting hands from effects of prolonged and repeated exposure to antiseptic solutions).
(3) Soak pieces of gauze swab in warm water and squeeze to remove excess water (too much water will dilute the agent, pool in body 'pockets' and risk the patient becoming cold during surgery).
(4) Apply several pumps of antiseptic solution on to swab.

(5) With firm action, begin surgical scrub: start at the incision site and work outwards towards the edge, taking care not to move swab from contaminated area to clean area.
(6) Lather the area well but avoid excessive scrubbing pressure as this will cause skin irritation.
(7) Discard each swab after reaching the periphery (do not return to the centre).
(8) Repeat the procedure using a clean swab each time.
(9) Continue until no more obvious dirt is removed by the swab.

Move patient to operating theatre.

(10) Continue scrub procedure as before (please note that some authorities advocate the use of sterile gloves and swabs for this part of the procedure but this will probably depend on the policy of the practice).
(11) Continue procedure to allow 5 minutes of antiseptic contact time.
(12) Alcohol may be applied after the scrub solution to remove the lather and provide additional antisepsis (do not use on open wounds, mucous membranes or small mammals susceptible to hypothermia).
(13) A final solution of povidone–iodine may then be used on the area using sterile swabs and Cheatle forceps. The discoloration of the skin alerts operating room personnel that the site has been prepared and should not be touched and the solution provides continued bactericidal activity.

DRAPING THE PATIENT

Drapes are one of the main barriers against contamination during surgery and should be large enough to cover the patient and the whole operating table. Drapes can be made of cotton, paper or plastic. Cotton drapes have the advantage of being reusable and therefore cost effective, and they conform well to the patient. However, their main disadvantage is that when wet, they become a carrier for bacterial strike-through from the patient to the surgeon. Water repellent paper drapes and plastic drapes reduce the risk of moisture contamination but can be more expensive.

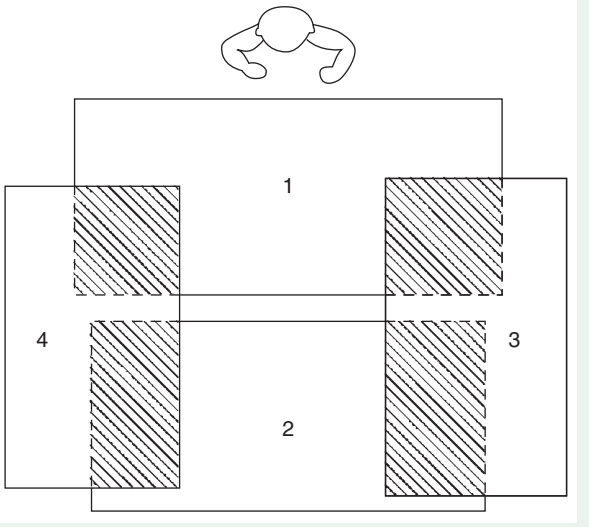
Draping should be carried out by a gowned and gloved member of the surgical team.

Four corner draping

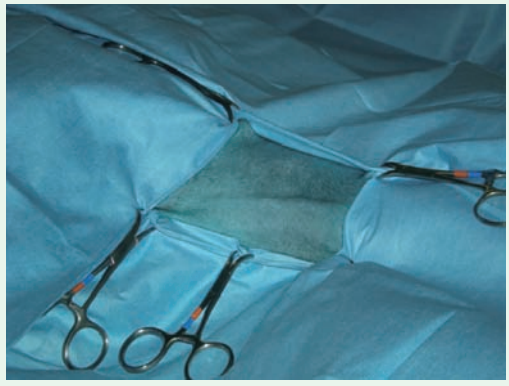
In this method of draping, four drapes are used to surround the proposed surgical incision site (Fig. 15.1). The patient and the entire table should be covered with the drapes to maintain an aseptic field and prevent the surgeon and assistant contaminating themselves or instruments during the surgical procedure.

Once placed, the drapes should not be moved towards the incision site to avoid bringing contaminants towards the site.

Fig. 15.1 Draping method.

Ask a non-scrubbed assistant to open drape packaging without touching the inside of the pack or its contents
Grasp the first drape firmly so it doesn't fall open and lift up and out of the packaging
Step away from any contaminating surfaces, such as the operating table or anaesthetic machine to allow sufficient space for the drape to open fully
Take hold of the two adjacent corners of the drape, one corner in between the thumb and forefinger of each hand
Allow drape to drop open
Open the drape fully by extending arms outwards whilst observing to ensure that the drape does not touch any contaminating surfaces
Create a double thickness of drape by folding over approximately 20 cm of the top of the drape away from yourself
Hold the drape at the folded corners and rotate the wrists inwards, wrapping the drape around the hands so that the hands are covered by the drape
Approach the table and lay the first drape between the patient and yourself (Fig. 15.1a)  15.1a
Lay this drape so that edge of drape lies parallel to and about 5 cm lateral to the incision site
Place one hand down at a time so that you are able to observe each hand whilst releasing the corners of the drape (this is to help minimise the risk of inadvertently touching any contaminated surfaces)
Once you have placed the drape, do not adjust drape towards the incision site

Retrieve second drape from packaging and open and fold over as before
Bring hands together (this will make drape smaller) and move around to opposite side of patient
Open up drape and lay second drape as before, opposite the first drape
Retrieve third drape from packaging, step aside and open and fold over as before
Walk around the patient and lay this drape caudally to cover the unclipped fur (include making the drape smaller whilst walking around to the patient)
Pick up towel clips and separate from each other
Attach first towel clip by placing the tips diagonally across one corner of the drapes
Pick up small fold of skin through the drape and close the clip
Tuck the towel clip under top drape
Attach second towel clip as above to the other corner of the drapes
Retrieve fourth drape from packaging and open and fold over as previous ones
Lay this drape cranially (you may need to 'fly' this drape across the patient if direct access to this side of the table is difficult (e.g. if the anaesthetic machine is in the way)
Attach final two towel clips in the remaining corners of the drapes and position under the drapes as before
Ensure all towel clips are attached to skin and drapes
Ensure that drapes cover all of the unclipped fur and that no 'long' fur is poking out of the fenestrated area (Fig. 15.1b)



15.1b

There may be occasions when it is advisable to add additional draping over the top to provide a second protective layer between the patient and surgeon. For example enterectomy or exploratory laparotomy with increased risk of contamination. In this case, adhesive plastic drapes may be useful. A large adhesive barrier drape is placed over the first layer of drapes, and directly on to the skin. The surgical incision is then made through this layer. These drapes provide a waterproof barrier and are quick to apply, however, the presence of stubble and moisture on the skin surface often prevents them sticking well.

The 'towelling-in' method of skin draping involves suturing disposable paper drapes to the skin edges after the initial skin incision is made. Whilst time-consuming, this provides an effective barrier against bacteria by completely isolating the surgical site from the surrounding skin surface.

Draping for orthopaedic/limb surgery

During any surgical procedure to the limb, it may be necessary to manipulate the limb and access various aspects of the leg. In this instance, a specific draping technique is useful. In this method of draping, a non-scrubbed member of the surgical team first applies a bandage or stockinette bandage over the distal (and often un-clipped) part of the limb. This is then either held upright, away from the body by the non-scrubbed assistant, or hung from a drip stand.

As before, the patient is draped using the four corner draping technique around the suspended limb.

(1) Ask the non-scrubbed assistant to hold the limb by the covered section (if the limb has been suspended from a drip stand, the assistant will first need to cut or remove the tape holding the limb).
(2) Select a small sterile drape and grasp the presented limb through the drape (taking care not to touch the non-scrubbed assistant).
(3) Carefully wrap the bandaged part of the limb with the drape.
(4) Secure with a towel clip.
(5) Cover the drape with sterile bandage and secure.

Chapter 16

Preparation of the Surgical Team

One of the possible sources of contaminants in the theatre is the personnel involved with the surgery. Changing into theatre clothing and scrupulous cleaning of the hands helps to reduce the risk. Table 16.1 lists the types of clothing necessary.

THE SURGICAL SCRUB

Effective surgical scrubbing of the surgeon's and assistant's hands and forearms is an essential part of the preparation to reduce the number of contaminants (dirt and transient and residual microorganisms) and because a high percentage of surgical gloves get holes in at some stage during the surgical procedure (Fig. 16.1).

The entire scrub should last between 5 and 7 minutes.

Choosing a skin antiseptic

As with the patient scrub, the considerations for selecting and using a particular type of antimicrobial skin scrub solution include:

- a detergent to remove gross dirt and grease;
- ability to remain active in the presence of organic matter;
- an antimicrobial agent effective against a wide range of microorganisms;
- long residual effect;
- causes no or minimal skin irritation.

See Chapter 15.

HAND DRYING

After the scrub procedure is completed, the hands are dried using a sterile towel (Fig. 16.2).

GOWNING AND GLOVING

Once your hands and forearms are dry, a sterile gown is then put on taking care not to touch the outside of the gown at any stage of the procedure or to contaminate the gown by touching non-sterile equipment or furniture.

The closed gowning technique (Fig. 16.3) is the preferred method when preparing for sterile surgical procedures. Open gloving technique should be used when only the hands need to be covered (i.e. not when using a gown).

The sterile gloves are put on immediately after putting on the gown. These can be put on using the closed (Fig. 16.4) or 'plunge' method.

Table 16.1 Theatre clothing.

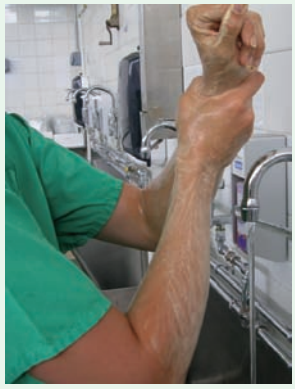
Attire	Notes
Scrub suit (top and trousers)	Comfortable, cool. Ensures outside clothes are not worn in theatre. Clean pair everyday or more often if necessary. Top should be tucked into trousers
Shoes/wellies	Comfortable, worn only in theatre. Clean regularly as will become blood splattered
Hats	Cloth or disposable – must contain all of hair. Hats are available with side covers for beards
Mask	Must cover nose and mouth – efficiency is reduced when moist
Sterile gown	Covers arms and body creating sterile area
Sterile gloves	Provides a sterile barrier to ensure asepsis

Fig. 16.1 Surgical scrub procedure.

Remove watches, bracelets and rings (nails should be short)
Turn on the taps to produce a steady flow of comfortable temperature of water
Keeping the hands higher than elbows, wet the hands and arms thoroughly
 Use one elbow to depress the antimicrobial soap dispenser and apply 2–3 pumps of antimicrobial soap on to free hand (Fig. 16.1a)

16.1a

Work the soap into a lather over both hands and forearms as far as the elbow (Fig. 16.1b)



16.1b

16.1c

Use an elbow to depress the lever on a wall-mounted scrub container to remove one scrub brush (or ask assistant to open brush container if different type used) (Fig. 16.1c)

Clean under nails with short nail-cleaning bristles of brush or nail stick, under running water (Fig. 16.1d)

16.1d

Rinse hands and forearms, still keeping the hands higher than elbows

Apply 2–3 pumps of antimicrobial soap – work into lather on hands and forearms up to the elbows

Note the starting time

Both hands: for approx. 4 minutes (or at least 10 strokes of the brush on each plane)

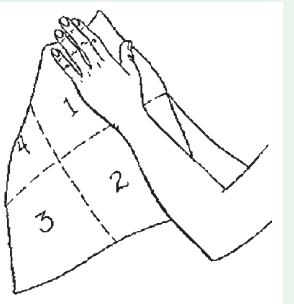
Scrub the nails

Scrub all four planes of each finger

Continued

Scrub the palm of the hand
Scrub the back of the hand
Use hands or brush to spread soap up forearm – two thirds of the way to the elbow
Repeat a short scrub procedure for each hand
Spread soap up the forearm – half way up to the elbow
Repeat a final short scrub procedure for each hand
Drop scrub brush into sink
Check clock to ensure that scrub procedure has taken no less than 5 minutes
Rinse both hands and arms thoroughly under the running water
Turn off the taps with elbows
Continue to keep hands higher than elbows and stand by sink to allow most of the water to run off into the sink
Ensure: Keep hands higher than elbows throughout Both hands scrubbed for appropriate length of time Do not touch any article with hands or forearm

Fig. 16.2 Hand drying technique.

Ask a non-scrubbed assistant to open the packaging containing the sterile towel
Remove the towel from pack taking care not to drip water on the gown beneath it
Keep your hands higher than your elbows whenever possible
Step back away from the table
Allow the towel to unfold
Hold a quarter of the towel over right hand and use this quarter to dry the fingers, palm and back of the left hand (Fig. 16.2a)  16.2a

Move the right hand down to the ‘second’ quarter (the one below the first) and dry the left forearm
Work from hand to the elbow – do not return to hand once the elbow is reached
Once the first hand and arm are dry, bring the dry hand to the opposite end of the towel
Hold the third quarter of the towel over left hand and dry your fingers, palm and back of your right hand
Move the left hand down to the last quarter of the towel
Use the last quarter of the towel to dry your right forearm
Work from your hand to elbow and do not return to hand once elbow reached (in the same way as before)
Drop the used towel into the receptacle provided or on to ‘dirty’ bench surface if receptacle not provided
Do not lower your hands below waist level throughout the procedure

Fig. 16.3 Closed gowning technique (using cotton gown).

Ask a non-scrubbed assistant to open the gown packaging, taking care not to touch the inside of the package or its contents
Grasp the whole gown firmly so it doesn’t fall open and lift it up and away from the table (Fig. 16.3a)  16.3a
Step away from the table to allow room for gowning

Continued

16.3b

Hold the gown by the neck and allow it to gently unfold away from you (Fig. 16.3b)

Guide one hand and arm through the sleeve of the gown (Fig. 16.3c)

16.3c

Do not allow your hand to push through the cuff of the gown

Repeat for the other arm

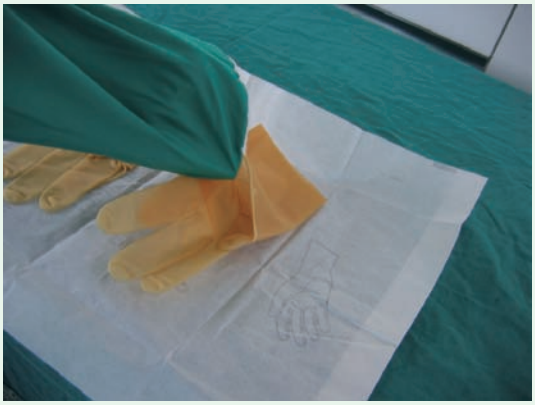
Ask the non-scrubbed assistant to pull the gown up over your shoulders by touching only the inside of the gown – and ask them to secure the ties at the back

Bend over slightly to pick up front or side waist ties

Hold the waist ties out to the side and ask assistant to take them (without touching you) and tie at the back

Ensure that throughout the procedure, the outside of gown is not touched by your hands, the assistant or any unsterile furniture or equipment

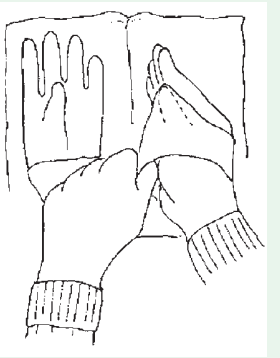
Fig. 16.4 Closed gloving technique.

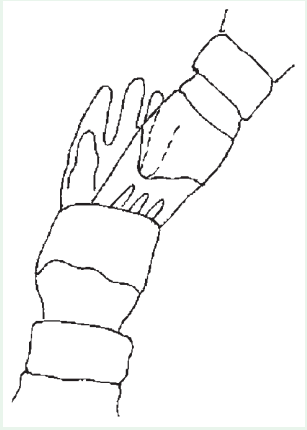
Keep your hands inside the sleeves of the gown
Ask the non-scrubbed assistant to open specified size of glove packet
Remove inner glove packet without touching outside of packet or assistant
Turn packet upside down and drop on to a sterile surface
Unfold packet to expose the gloves
With left hand, pick up the rim of the glove on the right (actually the left glove) (Fig. 16.4a)  16.4a
Turn the hand over so the palm is facing upwards – the glove is lying on the hand with the fingers still holding on to the rim of the glove (Fig. 16.4b)  16.4b
Using the right hand, grasp the other rim of the glove
In one smooth movement, pull the glove over the left hand until the fingers are in their correct places and the rim of the glove covers the gown cuff

Continued

With the right hand, pick up the rim of the glove on the left
Repeat the above procedure
Adjust the fingers and cuffs of both hands until comfortable
Clasp hands together and maintain higher than elbows

Fig. 16.5 Open gloving technique.

Push both hands through the cuff of the gown
Ask assistant to open specified size of glove packet
Remove inner glove packet without touching outside of packet or assistant
Drop the packet on to table top or bench
Unfold packet to expose the gloves
With left hand, pick up the folded edge of the glove on the right (Fig. 16.5a)  16.5a
Insert the fingers of the right hand inside the glove until the fingers are just in the fingers of the glove
Insert the thumb of the right hand inside the glove cuff (it is unlikely that you will be able to put your thumb inside the thumb of the glove at this stage)
Extend the thumb away from the fingers
Let go of the glove with the left hand

Insert the fingers of the gloved hand in between the cuff and the palm of the left glove (Fig. 16.5b)  16.5b
Insert the fingers of the left hand inside the glove until the fingers are just in the fingers of the glove
With one smooth movement, pull the cuff of the left glove up and over the cuff of the gown
Using the left (gloved) hand, now insert your fingers in between the cuff and the palm of the right hand
With one smooth movement, pull the cuff of the right glove up and over the cuff of the gown
Adjust fingers of each glove so the gloves are comfortable and fitted
Ensure that both cuffs of the gown are included under the cuffs of the gloves
Clasp hands together and maintain higher than elbows
Maintain sterility of self throughout procedure

The plunge gloving technique is the easiest of all methods. A scrubbed and gloved assistant holds the gloves open for the surgeon to 'plunge' their hand straight into the glove. Ensure the assistant knows their left from their right!

Open gloving (Fig. 16.5) should not be used routinely for preparation for surgical procedures. This method cannot provide as much of an aseptic barrier for microbes and should only be used when the hands are to be gloved, for example, during bandage and dressing changes, urinary catheterisation, intravenous catheter placement.

Chapter 17

Assisting During Surgery

INTRAOPERATIVE CARE – THE SCRUB NURSE

The VN is an important member of the surgical team and must be fully competent in assisting the veterinary surgeon during surgical procedures. A good assistant is invaluable and will help speed up the surgery and therefore reduce anaesthetic time.

The main tasks of the scrub nurse during a surgical procedure include:

- assisting the surgeon (without distracting or slowing the surgeon down);
- assisting with the control of haemorrhage;
- anticipating the surgeon's needs;
- keeping the instrument trolley tidy;
- handling body tissues and keeping them moist;
- cutting sutures correctly.

It is important to note the following points when assisting with surgery.

(1) When assisting during a surgical procedure, take care not to block the surgeon's view of the operating field or create shadows by standing in the way of light sources.
(2) Know all the names of the instruments that are being used during each procedure and anticipate which instrument will be required next.
(3) Pass the instrument to the surgeon so that the handle is placed into their hand: take particular care when passing scalpel blades and sharp instruments.
(4) Keep the instrument trolley in order. At the start of surgery, arrange instruments on the trolley in the anticipated order of use (e.g. scalpel handle, scissors, rat tooth forceps etc.).
(5) Place used, bloody instruments on one side of the trolley and keep them separate from the clean, unused instruments.
(6) Control haemorrhage throughout surgery using gauze swabs as necessary. Anticipate when you will run out of gauze swabs and request more from the theatre nurse.
(7) Count all swabs at the start and end of each procedure. Keep track of the number used to avoid any being accidentally left inside a body cavity.
(8) When swabbing haemorrhaging tissues, use a dabbing motion rather than wiping as this can impair clotting and damage delicate tissue.

(9) Control haemorrhage from a vessel by applying pressure with a finger or gauze swab until the surgeon can apply a haemostat (the tips of the clamp trap the severed ends of the vessel). This may be left in place or ligated with suture material.

(10) Pass cautery instrumentation for haemorrhage control to the surgeon if it is available.

(11) Moisten sterile gauze swabs with warm saline and apply to any drying tissues. Drying can lead to tissue damage.

(12) If suction equipment is available and necessary (e.g. during abdominal lavage or severe haemorrhage) use an appropriate suction tip and try to avoid adhering the end of the tip to the surrounding soft tissues.

(13) Handle all tissues and organs gently; aggressive manipulation of these can cause trauma, bruising and haemorrhage.

(14) Do not replace bloody swabs or discarded tissue matter on to the trolley, drop them into the kick bin.

(15) Cut sutures to the required length after a knot has been tied. Use blunt-ended scissors and avoid inadvertently nipping any surrounding tissues.

(16) Be aware of maintaining aseptic technique at all times throughout the procedure. If asepsis is accidentally broken, tell the surgeon and reglove.

(17) Remember to breathe! Many surgical assistants feel faint even having worked in the theatre for years. This is probably due to the fact that the assistant often has to stand very still in one position for long periods, concentrating hard whilst holding a particular body part. Adjusting bodyweight from one foot to the other is also important to keep the circulation moving.

Chapter 18

Skin Suturing Techniques

SUTURE MATERIALS

Characteristics of suture material

- Knot security: how many 'throws' are required.
- Capillary action: its ability to suck up fluid along its length.
- Tissue drag: friction that occurs when throwing a knot.
- Memory: ability to retain shape.
- Chatter: friction caused when it passes over itself.
- Sterilisation: how it can be sterilised.
- Tissue reaction: irritant effect on tissues.
- Elongation: stretchiness before breaking.
- Tensile strength: strength before it breaks.

Classification of suture materials

In the past many different materials were used to make sutures, including intestinal tissue and kangaroo tendons. These days, however, the most commonly used materials are synthetic, although there are still a few natural products in use such as gut and silk.

A suture is classified in three ways depending on what it is made of (Table 18.1):

- absorbable or non-absorbable;
- natural or synthetic material;
- monofilament or multifilament.

Alternatives to suture materials

Staples

Staples may be used for skin wound closure, for lung lobectomies, liver biopsies and bowel resection. They are expensive and need special equipment for the application of the staples. The main benefit is that they are extremely quick and provide good closure when used in internal organs.

Adhesive tapes

These are available for wound closure but they are expensive and do not stick well to moist skin or skin with animal hair stubble.

Table 18.1 Suture material classification.

Classification	Details
Absorbable	Lose their tensile strength between 10 and 40 days Are totally absorbed between 40 and 180 days Are absorbed by either phagocytosis or hydrolysis Are used for wound closure where long-term support is not needed
Non-absorbable	Maintain their tensile strength >60 days Become encapsulated in tissue Can be used where prolonged mechanical support is required, e.g. skin closure and slow-healing tissues
Natural	Made from natural fibres, e.g. catgut (sheep or goat intestines), linen Natural absorbable sutures are removed by phagocytosis and produce some tissue reaction
Synthetic	Less reactive, more reliable action Synthetic absorbable sutures are removed by hydrolysis (broken down by fluid) and so there is minimal tissue reaction
Monofilament	Single strand material, reduces tissue drag but can be difficult to handle
Multifilament	Multistrand fibre which is either braided or twisted. Adds strength but will increase tissue drag unless coated

Tissue glue

Cyanoacrylate monomers are used to glue skin edges together, but they are toxic and have been shown to cause a granuloma reaction.

Selection of suture materials

Examples of appropriate materials for particular body tissues.

- ***Skin:*** monofilament nylon, polypropylene.
- ***Subcutis:*** fine, synthetic, absorbable, e.g. Vicryl.
- ***Fascia:*** synthetic, non-absorbable (nylon, polypropylene) or absorbable such as PDS, Vicryl.
- ***Muscle:*** synthetic absorbable or non-absorbable nylon or polypropylene for cardiac muscle.
- ***Hollow viscus:*** synthetic absorbable or polypropylene.
- ***Tendon:*** stainless steel, nylon, polypropylene.
- ***Blood vessels:*** polypropylene.
- ***Nerves:*** nylon or polypropylene.

Tissue healing rates

When selecting an appropriate suture material for use in a particular body area, it is useful to consider the tissue healing rates of that area so that the most appropriate suture material can be selected (Table 18.2).

Table 18.2 Tissue healing rates.

Tissue	Time for healing
Skin	7–10 days
Fat	5 days
Muscle	14 days
Fascia	42 days
Serosa/mucosa	2–3 days

Table 18.3 Suture sizes.

USP	Metric
5–0	1
4–0	1.5
3–0	2
2–0	3
0	3.5
1	4
2	5
3	6

Selection of size of suture material

When selecting the size of the suture material, the VN should always aim to use the smallest diameter possible for each particular body part and the size of the animal.

The size of suture material can be expressed using two different methods (Table 18.3):

- metric;
- USP (US Pharmacopoeia).

The metric figure refers to the diameter of the suture material in 0.1 mm. For example:

- actual size = 0.4 mm;
- Metric size = 4 (multiply actual size by 10).

SUTURE NEEDLES

Suture needles are made from stainless steel, occasionally coated with silicone to facilitate passage through the tissues. The method of attaching the suture material to the needle may be:

- through an 'eye';
- swaged.

Eyed needles:

- are reusable;
- can be used with any type or size suture material;
- are cheaper;
- are more bulky to go through tissues;
- however, repeated sterilisation dulls cutting edges.

Single-eye needles should never be double-threaded as the bulk of the suture material causes severe tissue drag.

In swaged needles the suture material is attached directly. They:

- are traumatic to tissues;
- are expensive;
- are sharper;
- have ensured sterility.

Shape of needles

Suture needles come in a variety of shapes:

- straight (used for hand suturing – not to be used with needle holders);
- half-curved;
- curved.

The choice of needle shape is usually governed by the accessibility of the tissue to be sutured, and normally the more confined the operative site the greater the curvature required.

Needle points

- ***Cutting*** needles can be used in very dense or tough tissues. They are designed to be used in the skin. They incise a hole larger than the needle shank, therefore reducing tissue drag.
- ***Tapercut*** needles have a cutting tip on the point and a round body. They do not cut but spread the tissue and cause very little tissue trauma. They can be used in any type of tissue.
- ***Round-bodied*** needles do not cut – they are designed to separate tissue fibres rather than cut them. They are used for non-fibrous tissue, soft tissue organs and any delicate tissue where the tissue fibres are split easily.
- ***Reverse cutting*** needles look similar to cutting needles, but the difference is that the apex cutting edge is on the outside of the needle curvature. This improves the strength of the needle and increases its resistance to bending.
- ***Spatulated*** needles are extremely fine with very sharp cutting edges. They are used primarily for ophthalmic work.

Needle size and strength

Needle sizes come in a range from 4 to 24. Higher numbers indicate a finer needle. The diameter of the needle is a major factor in determining its strength, although the cross-sectional shape and type of wire are also important. The size of needle should be selected taking into consideration the tissue to be sutured, the size of the suture material required and the force required to push the suture needle through the tissue.

Most needles are designed so that they bend rather than break when they are over-stressed. This bending indicates that the needle has been used in a situation where a force has been applied which is greater than that for which it was designed. If bending occurs, the needle should be discarded rather than any attempt made to straighten it.

Use of needle holders

- The needle holder should be carefully selected to match the size of the needle being used. An overly large needle holder can cause damage and bend the needle.
- The needle size and curvature should match the size of the tissue bite required. Use of too small a needle for a given tissue will lead to bending of the needle.
- The needle holder should be in good condition – worn jaws result in needle rotation and instability of the needle.
- The needle should be held only on the flattened area and should not be grasped near the needle point or attachment or eye area. Non-flattened needles should be grasped at approximately one third of the total needle length from the suture material end.
- The force required to pass the needle through the tissues should be applied in the direction of the curvature of the needle.
- The needle should be inserted into each side of the tissue separately and should not be used to bridge a wound.

SUTURE PATTERNS

The suture pattern used in any area depends on the wound tension and personal preference and expertise. Suture patterns can be divided into:

- interrupted;
- continuous.

They are further classified into:

- simple;
- mattress;
- tension.

The most commonly used patterns are described below.

Simple interrupted

This is used in many areas of the body. The sutures are easy to insert and maintain good tissue apposition (Fig. 18.1 and Fig. 18.2). Occasionally, the wound edges may invert or if the sutures are placed too far apart, the wound will gape in between the sutures.

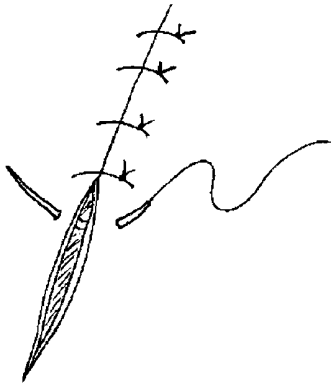
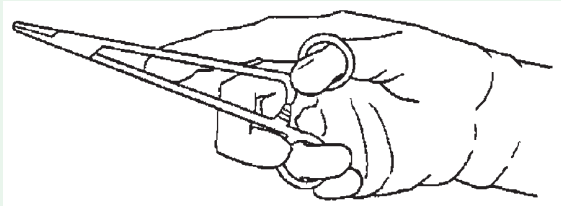
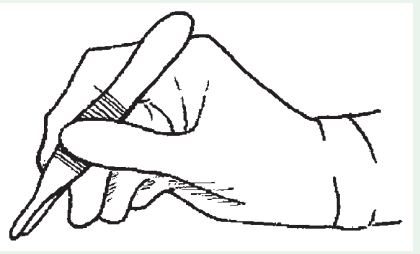


Fig. 18.1 Simple interrupted suture pattern.

Fig. 18.2 Simple interrupted suture placement.

Select appropriate suture material (e.g. monofilament nylon, e.g. 3 metric Supramid™)
Select the most appropriate needle for this wound (e.g. 16 mm curved, cutting needle)
Thread needle – do not double tread the suture material through the eye of the needle or tie a knot
Select appropriate needle holder (Mayo-Hegar)
Hold needle holder using thumb-ring finger grip in leading hand (Fig. 18.2a)  18.2a
Pick up needle and suture material and grasp needle with needle holders: • Position needle at right angles to the needle holder • Grasp the needle at half or two thirds of its length • Grasp in first third of the jaws of the needle holders
Lock needle holders on first ratchet

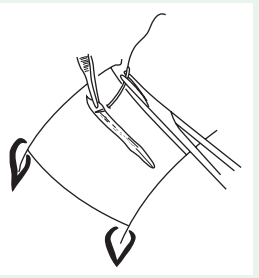
Continued



Hold rat tooth forceps in other hand, in pencil grip (Fig. 18.2b)

18.2b

At one end of the wound, stabilise the skin on the wound's far edge with rat tooth forceps



Push needle completely through the skin:
- From the far edge of the wound towards you
- Approx. 5–7 mm away from the skin edge (Fig. 18.2c)

18.2c

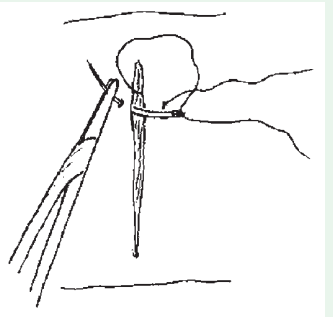
Release needle and skin from both instruments

Re-grasp the tip of the needle and pull suture through the skin leaving approximately 3 cm of free end

Stabilise the skin on the wound's near edge with rat tooth forceps

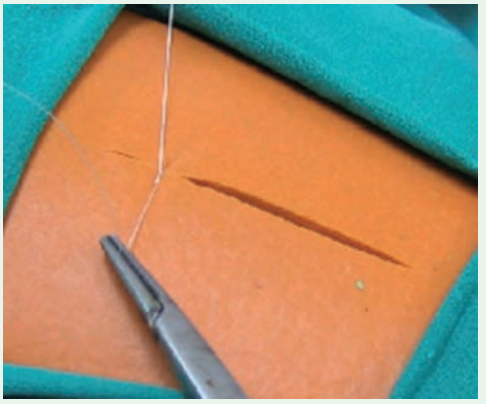
Push the needle through the underside of skin 2–3 mm away from the skin edge

Release the needle and skin



Re-grasp the tip of the needle and pull needle through the skin (Fig. 18.2d)

18.2d

Surgeon's knot tying technique
Palm the rat tooth forceps and gather up long end of suture material and needle in same hand
First throw: place the needle holders in between the two strands of suture material over the wound
Wrap the long end of suture (nearest you) twice around the end of the needle holders
Grasp free end (short end) of suture in needle holder tips
Bring short end towards you through the loop
Both hands cross over wound
Tighten suture gently over the wound (Fig. 18.2e)  18.2e
Release needle holders
Place the needle holders in between the two strands of suture material over the first knot
Second throw: wrap the long suture strand (furthest away from you) around the end of the needle holders
Grasp and pass the short end through the loop away from you
Both hands cross over the wound
Keep the hands low to ensure square knot is formed
Third throw is performed in the same way as the first throw and tightened down to secure the knot
Ensure even tension is applied when tightening sutures (to avoid half-hitch forming)
Cut suture leaving approximately 1–1.5 cm
Wound edge is held together with good tension (i.e. not too tight or too loose)

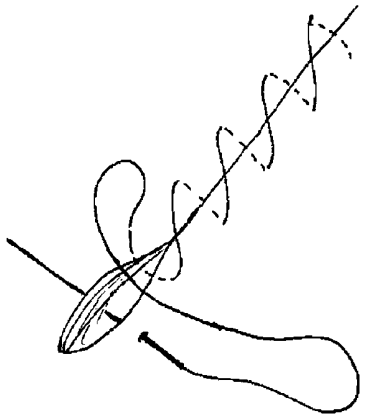


Fig. 18.3 Simple continuous suture pattern.

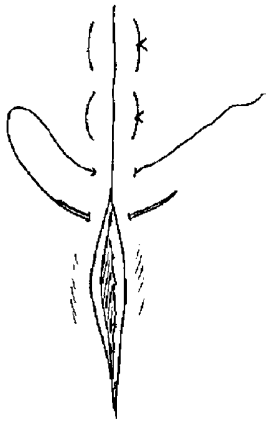


Fig. 18.4 Horizontal mattress suture pattern.

Simple continuous

This is often used to close subcutaneous tissues and skin. It is extremely quick to apply and gives good skin apposition. The major disadvantage with this technique is that a break in the suture line or knot will lead to total relaxation of the suture line and wound breakdown (Fig. 18.3).

Horizontal mattress

This is used for skin closure, especially when the edges of the wound are under a certain amount of tension (Fig. 18.4).

Vertical mattress

See Fig. 18.5.

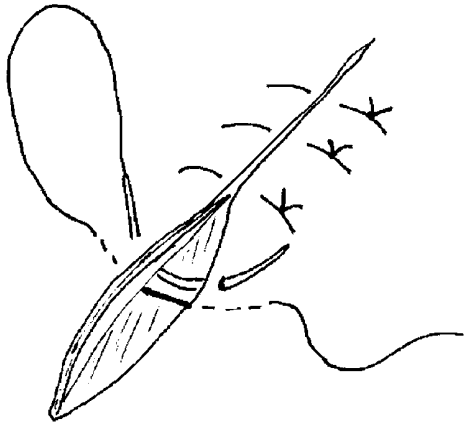


Fig. 18.5 Vertical mattress suture pattern.

Cruciate mattress

This is used for skin closure and provides good apposition of the wound edges (Fig. 18.6). It is useful when there is moderate tension and is quicker than using simple interrupted sutures. Removal is quicker as well.

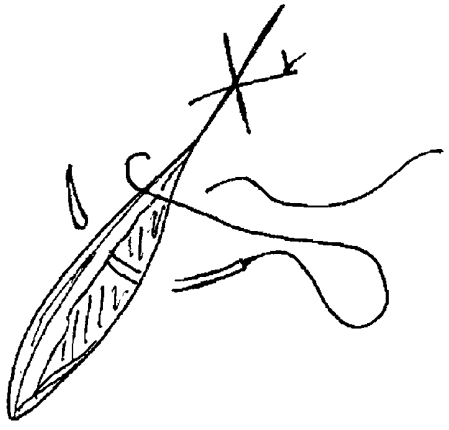


Fig. 18.6 Cruciate suture pattern.

Purse string suture

This is used to temporarily close an orifice (e.g. anus during surgery around that area). It is quick to apply and remove.

Chapter 19

Practical Care of Wounds

There are four main factors to consider in the management of wounds:

- the type of wound (classification);
- initial treatment and care of wounds when first seen;
- continued management of wound with appropriate dressing materials;
- signs of wound breakdown and delayed healing.

CLASSIFICATION OF WOUNDS

- Clean: a surgical wound made under aseptic conditions. There is no break into a contaminated area such as the respiratory or gastrointestinal tract.
- Clean-contaminated: another surgical wound, made under aseptic theatre conditions, but where it incorporates an incision into a contaminated area such as the respiratory, gastrointestinal or urogenital tract. However, there has been no spillage of the contents of the contaminated area into the wound.
- Contaminated: as above but spillage has occurred from one of these contaminated areas into the wound. Also may occur in traumatic injury such as one immediately following a road traffic accident (RTA) or animal bite, where contaminants are present but not sepsis (yet).
- Dirty: an infected wound. Either from a surgical wound into an area of the body containing bacteria such as the gut or from a contaminated traumatic wound over 6–8 hours old.

Contaminated wounds can become dirty wounds if the debris is not removed within 6–8 hours. This is called the 'golden period'. This refers to the time it takes for bacteria to be implanted somewhere and then begin to grow and multiply. This is why it is so important to thoroughly flush and clean all wounds as soon as possible, rather than leaving them until later.

INITIAL TREATMENT AND CARE OF WOUNDS

Wound healing will not occur until debris and infections are removed. Cleaning and decontamination of the wound is the most effective way of reducing long-term bacterial contamination and should ideally be carried out in the first 6–8 hours after injury. Aggressive action should be taken to thoroughly lavage and debride the wound. Ideally, this should be carried out in a clean/sterile environment (e.g. theatre or theatre preparation room).

Wound cleaning

(1) If the wound is older than 8 hours, obtain a swab for bacterial culture and sensitivity.
(2) Apply a water-soluble lubricating gel to the wound. This makes it easier for the clipped fur to be removed from the wound during lavage.
(3) Clip hair from around the wound. Use scissors for clipping the hair around the wound edges. It may help to wet the scissors with lubricating gel so that clipped hair sticks to the scissors rather than falling into the wound.
(4) Lavage the entire wound with copious quantities of warmed sterile, isotonic saline or Hartmann's.
(5) If the wound is very contaminated, warm tap water may be used initially to remove gross dirt, however, prolonged used of tap water will damage the exposed tissues because of its osmolality.
(6) Some people advocate the use of mild antiseptic solutions (2% povidone–iodine) but many antiseptic solutions are in fact an irritant to the tissues and their use has not been shown to be of any additional benefit to the long-term wound-healing process.
(7) Apply appropriate sterile dressing material and bandage.

Lavage technique

Use of a 20 or 30 ml syringe and 19 gauge hypodermic needle to lavage the wound has been shown to provide the ideal lavage pressure. High pressures run the risk of forcing debris and bacteria further into the wound and low pressures have been shown to provide ineffective lavage. Aerosol pumps, attached to drip bags, are commercially available and provide optimum lavage pressures.

Debridement technique

Debridement may be carried out on necrotic tissue using a sterile scalpel blade, scissors and forceps. This must be carried out in a sterile theatre environment. Layered, staged and en-bloc resection are methods of debridement and should only be carried out following training and instruction from the veterinary surgeon.

CONTINUED MANAGEMENT OF WOUNDS USING DRESSINGS AND BANDAGES

After the trauma of a wound, the fluid that is immediately released into the area dries out and forms a scab. A scab is the body's natural wound dressing, designed to protect the healing wound and prevent further contamination. However, a scab will delay the healing process in two ways: it increases the amount of necrotic material within the wound and also means that the migrating epithelial cells have to force a route beneath the scab. Wounds that heal in this way are far more likely to scar.

The aim of wound management is to prevent the wound from drying out and prevent the formation of a scab. This allows the epithelial cells to migrate rapidly over the wound surface.

Dressings

There are many ways to classify wound-dressing materials. For example, the following terms are used:

- dry dressings;
- moist dressings;
- wet dressings;
- impregnated gauze;
- adherent or non-adherent dressings.

However, this is not really very important. What is important is that the VN knows what is available and what is most appropriate for a particular wound at each stage of its healing process.

Dry dressings (non-adherent)

Generally, dry dressings such as Melolin™ may be used on surgical wounds where the edges of the wound are held together. For most other types of wound, they have been shown to be detrimental to wound healing. They tend to adhere to the wound surface, causing pain and disruption of the healing processes upon removal.

Dry-to-dry

The method of dry-to-dry dressings has gone out of fashion. Their main application is for dirty, necrotic wounds that have lots of loose tissue and foreign matter to remove. Sterile surgical gauze is applied dry to the wound surface. When it is removed (each day), a layer of tissue is removed along with the necrotic material and debris.

The main reason for dry-to-dry dressings no longer being used is that removal can be extremely uncomfortable if not painful for the patient and it is not just the necrotic tissue and debris that are removed. A certain amount of the healthy, healing cells are disrupted and removed. This type of dressing should never be used in wounds with good granulation and epithelialisation.

Wet-to-dry

Wet-to-dry dressings work in a very similar way to dry-to-dry dressings. The gauze swab is first soaked in sterile saline before being applied to the wound. This reduces the viscosity of wound exudate so that it may be absorbed more easily. The fluid then evaporates and the dressing becomes dry by the time it is removed, thus pulling away everything else with it.

These dressings have added disadvantages over dry-to-dry dressings because the moist environment is conducive to bacterial growth and also the

tissues surrounding the wound may become macerated if the dressing is too wet. The use of cold wetting solutions can cause discomfort to the animal.

Impregnated gauze dressings

These are slightly less adherent than dry dressings but they do very little to actively promote wound healing. Examples include Jelonet™, Grassolind™.

Poultice

A poultice is a paste or impregnated dressing that is applied to an area to draw out infection; they are used more widely in large animal medicine than in small animals. Animalintex™ is the most commonly used commercial poultice.

Moist dressings (non-adherent)

With sophisticated materials available commercially, there is no need to continue using the outdated methods described above. Moist dressings are now the materials of choice.

Moist wound dressings (hydrogels such as Intrasite™ and Biodress™ and hydrophilics such as Allevyn™) are interactive dressing materials that are extremely useful in maintaining a moist wound environment. Their use has been shown to accelerate wound healing by providing an optimum wound temperature, encouraging the migration of cells from the edges of the wound and encouraging cell mitosis. They are able to remove excess exudate and toxic components whilst giving protection from secondary infection. They are easy to remove and do not cause trauma or pain at dressing change.

Note: There is no single dressing material that can provide the optimum environment for all wounds or for the total healing stages of one wound, therefore it is important to select the most appropriate dressing for each wound at each particular stage of the healing process. This is why it is important for the VN to research available products and keep up to date with new materials.

Bandages

A VN should be proficient in all bandaging techniques ensuring that appropriate dressings are used. New products are always being developed as are new techniques for wound management.

A bandage should consist of three layers; each layer having its own distinct characteristics and functions.

- Primary dressing (contact layer):
  - sterile;
  - must remain in close contact with the wound;
  - must allow drainage of exudate;
  - must prevent contamination from environment.
  - Examples: Melolin™, Intrasite Gel™, Allevyn™, Rondopad™.
- Secondary dressing (intermediate padding layer):
  - absorbs fluids;
  - provides padding;

  - provides comfort and immobilisation.
  - Examples: Cotton wool, Soffban™, Ortho-band™.
- Tertiary dressing (outer layer):
  - secures other dressings/bandages in place;
  - provides additional support and protection;
  - protects other dressings/bandages from getting dirty.
  - Examples: Cohesive bandages: Co-Form™, Co-Flex™, Powerflex™. Adhesive bandages: Bandesive™, E-Band™, Elastoplast™, Treatplast™.

Rules of bandaging

(1) Collect all necessary equipment first.
(2) Ensure the animal is adequately restrained.
(3) Always wash hands prior to handling and wear sterile gloves if necessary.
(4) Apply layers evenly, prevent areas of excess pressure (hocks, elbows).
(5) Do not unroll too much bandage at a time.
(6) A bandage on a swollen limb will become loose once swelling has subsided.
(7) Avoid sticking too much adhesive to fur (remove with alcohol).
(8) For fractures: include proximal and distal joints to adequately immobilise the fracture.
(9) Check that the bandage is not too tight: it should be possible to insert two fingers between the bandage and the animal.
(10) Change dressings as often as necessary, and check frequently.

The VN should wear gloves and a protective apron to maintain personal hygiene and health when changing any dressing.

Prevention of self-trauma

Animals may try to remove a dressing or bandage if it is uncomfortable or painful. Check if this is the problem before trying methods of prevention, which include:

- Elizabethan (Buster™) collars;
- neck braces (Bite Not™);
- bandages (or old T-shirts, socks, etc.);
- topical applications (Bitter apple or Tabasco sauce);
- sedation (last resort).

Pressure bandages

These are used to control haemorrhage and prevent excess swelling and oedema. A pressure bandage should be removed after 12–24 hours, as it can reduce blood supply to the tissues. It should also be changed if the animal appears uncomfortable. The VN should avoid putting excess padding on convex surfaces, as this increases the pressure in this area. Pressure bandages must be marked so everyone is aware of them and knows when they should be removed. Using colour coded bandages will help identification.

After-care of bandages and client information

It must be stressed to owners (and hospital lay staff) that the success of treatment depends on good care of the animal's dressing and bandage. The VN or client must inform the veterinary surgeon if any of the following occurs:

- bandage begins to smell;
- bandage slips from original position;
- areas of soreness develop around the bandage;
- discharge seeps through the bandage;
- persistent patient interference.

It is important to keep the bandage both clean and dry. When the animal is being taken outside, the lower part of the bandage should be covered with a plastic bag or empty drip bag and secured with a piece of tape or loose elastic band. The bag must be removed as soon as the pet is back inside or the foot will sweat.

Bandaging techniques

Useful tips for bandage application:

- 'Stirrups' will help prevent limb bandages slipping down the limb and are useful for most limb bandages (the tapes should not be applied over wounds or areas with skin lesions).
- Avoid sticking adhesive bandages to the fur if possible. They pull the hairs causing discomfort, which frequently results in the patient chewing or interfering with the bandage.
- Always unroll bandage as shown in Fig. 19.1. This prevents the operator running out of bandage.
- When bandaging a limb, start distally and work proximally, rather than working down the limb.

Forelimb/hindlimb bandage

This is used to hold wound dressings in position, postoperatively for support and to help control inflammation, and as a first aid support measure for fracture management (Fig. 19.1).

- Check length of claws, clip if necessary.
- Cover any wounds with an appropriate dressing.
- Cotton wool between toes and pads, do not forget the dew claw.
- Position joints in natural angles, do not over-extend limb.
- Apply synthetic padding bandage layer or cotton wool around the limb.
- Apply splint if required.
- Apply conforming bandage to the distal limb first before winding evenly up the limb, overlapping the previous layer by half the bandage width.
- Separate distal ends of the tape strips and turn over and extend up along the proximal bandage to adhere to the conforming layer.
- Place protective layer of cohesive bandage or adhesive bandage over the top.

Fig. 19.1 Applying a forelimb bandage.

Select and collect all of the following equipment *before* continuing

Dressing material (Fig. 19.1a)

19.1a

Cotton wool

Padding layer (Fig. 19.1b)

19.1b

19.1c

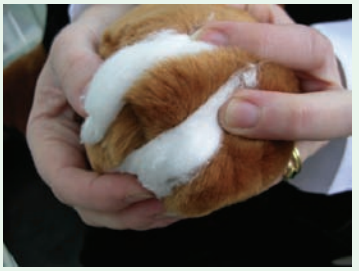
Conforming layer (Fig. 19.1c)

Protective outer layer (e.g. Bandhesive™) (Fig. 19.1d)

19.1d

Wash hands

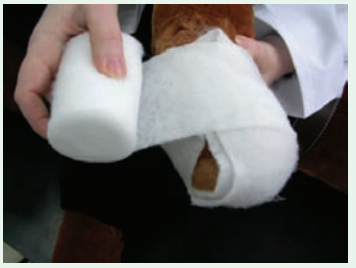
Apply small pledgets of cotton wool between the digits, the metacarpal pad and digital pads (optional) (Fig. 19.1e)



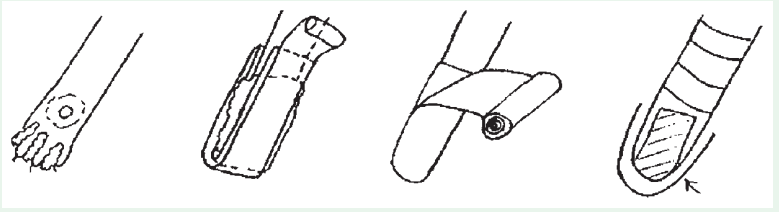
19.1e

Apply wound dressing – taking care not to touch the contact surface of the dressing (shiniest side to the wound)

Wrap padding layer around foot – this can either be done as shown below or in a figure of eight pattern (Fig.19.1f and Fig. 19.1g)

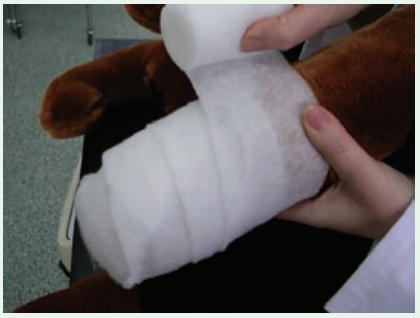


19.1f



19.1g

Start at distal end of foot – level with 2nd and 5th digits



19.1h

Wrap obliquely and overlap half of the width of the bandage (Fig. 19.1h)

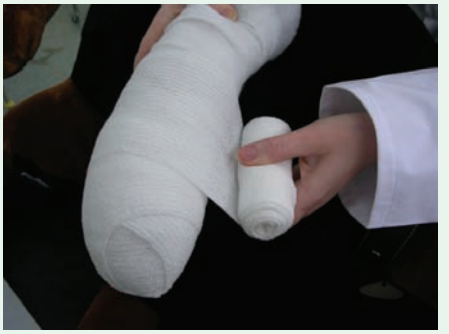
Continue up the limb – above the elbow

Continued

Use sufficient padding (i.e. several layers)

Apply conforming bandage layer in the same way as above

From distal to proximal limb (Fig. 19.1i)



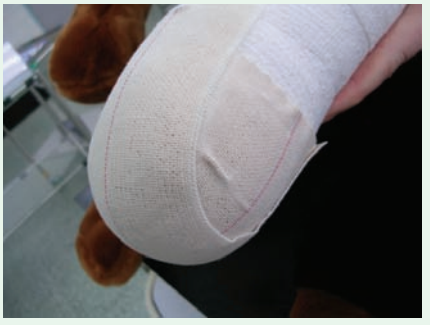
19.1i

Overlapping half the width of the bandage

Use appropriate tension to ensure bandage provides protection without overstretching and becoming too tight

Cut two short lengths of protective layer

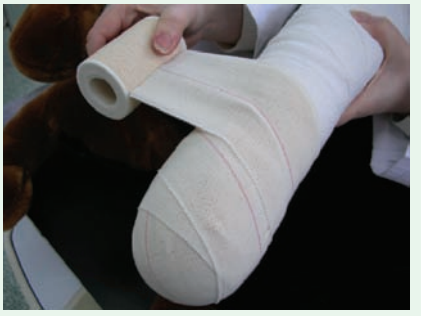
Apply first piece over distal end of foot – cranial to palmar aspects



19.1j

Apply second piece over distal end of foot – lateral to medial (Fig. 19.1j)

Wrap protective layer over entire bandage (Fig. 19.1k). Work from distal end to proximal. Limb is now bandaged correctly – neat and tidy with no bits of cotton wool sticking out



19.1k

Ensure bandage is not overly loose or too tight

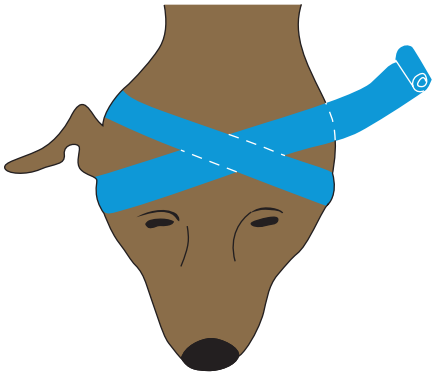


Fig. 19.2 Head bandage.

Ear and head bandage

This is used to control haemorrhage from pinnae, following aural resection and surgery for aural haematoma.

- Apply dressing to wound.
- Place pad of cotton wool on top of head.
- Fold ear back on to pad and cover with further padding.
- Apply conforming bandage over the top, passing either side of the free ear in a figure-of-eight pattern (Fig. 19.2).
- Cover with cohesive outer layer.
- Make sure that the bandage layers are not applied too tightly. It should be possible to slip two fingers underneath the bandage easily and the patient should be able to breathe easily and open its mouth.
- Draw over the top with felt-tipped pen the position of the covered ear to prevent cutting through the pinna when removing the bandage.

Abdominal bandage

This is used following abdominal surgery or trauma, to hold a wound drain in place or to hold a gastrostomy tube in place (Fig. 19.3).

- Cover wound or incision with dressing.
- Use minimal padding – enough for comfort but too much will cause the bandage to slip.
- Use wide conforming bandage followed by a cohesive bandage (Elastoplast is too restrictive).
- You may include the forelegs or hindlegs to prevent bandage slipping or bunching up.

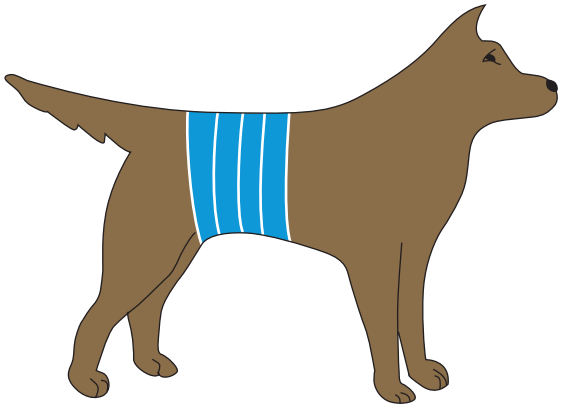


Fig. 19.3 Abdominal bandage.

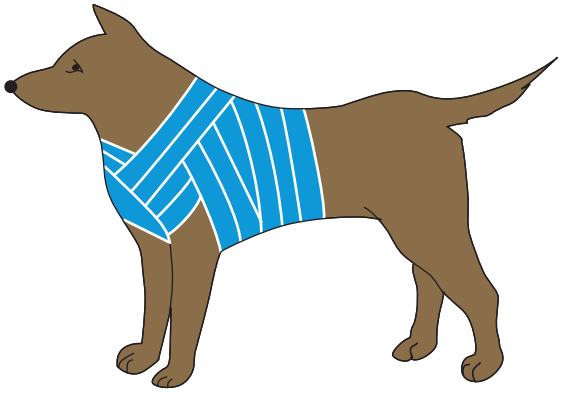


Fig. 19.4 Thoracic bandage.

Thoracic bandage

This is used following surgery or trauma, and to retain a chest drain.

- Cover wound or incision with dressing.
- Use minimal padding.
- Use wide conforming bandage.
- Cover with cohesive bandage not elastic adhesive.
- Include forelegs in figure-of-eight fashion to anchor bandage (Fig. 19.4).

Tail bandage

A tail bandage is used following tip amputation to prevent further trauma.

- May use similar method to limb bandage, but with less padding
- Stent dressing: swab is sutured to end of tail.
- Syringe case is adapted to fit over the end and then secured to the tail.

Robert Jones bandage

This is used as a first aid support, to immobilise a fractured limb and to give additional support following joint surgery (Fig. 19.5).

- Apply traction tapes to dorsal and ventral surfaces of lower limb.
- Unroll cotton wool from roll and wrap firmly around the limb.
- Cover first padding layer with conforming bandage.

Fig. 19.5 Applying a Robert Jones bandage.

Select and collect all of the following equipment *before* continuing

2 × zinc oxide tape strips cut to approximately 30 cm (for stirrups) (Fig. 19.5a)

19.5a

Cotton wool – 1 roll

19.5b

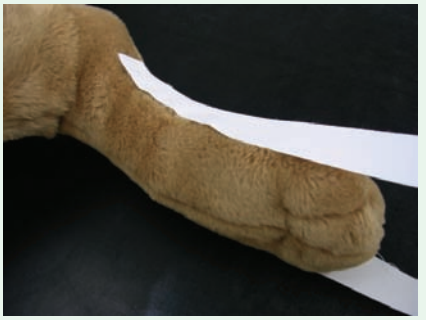
Conforming layer (2–3 packets) (Fig. 19.5b)

Protective outer layer (e.g. Bandesive™) (Fig. 19.5c)

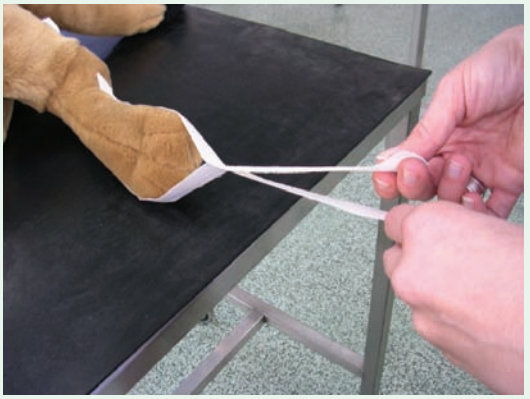
19.5c

Continued

Apply stirrups – one third of the length of the tapes to either the cranial and caudal aspect of the distal limb or to lateral and medial aspect of the distal limb (Fig. 19.5d)



19.5d



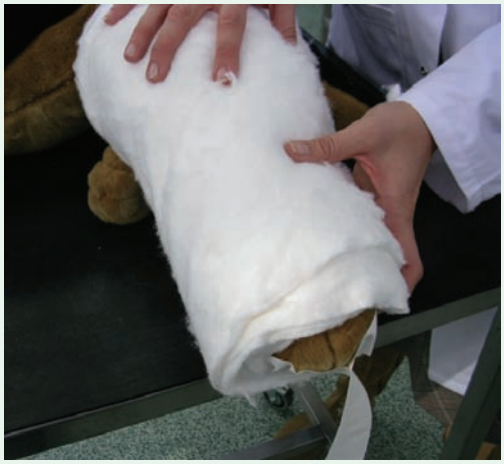
19.5e

Lightly stick the free ends of the stirrups together to prevent them sticking to the bandage material (Fig. 19.5e)

Wrap cotton wool padding layer around limb, leaving the two central toes exposed (Fig. 19.5f)

19.5f

Ensure this goes to above the stifle



19.5g

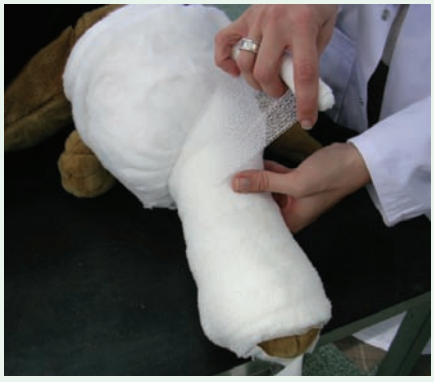
Use sufficient padding (i.e. wrap at least four layers) (Fig. 19.5g)

Apply conforming bandage layer (Fig. 19.5h)

19.5h

Leave stirrups hanging out from the bottom of the bandage (ask assistant to hold out of the way if necessary)

Wrap conforming layer tightly to compress the cotton wool layer (Fig. 19.5i)



19.5i

Continued

Apply from distal to proximal limb overlapping half the width of the bandage
Use appropriate tension to ensure bandage provides high level of support
Separate the pieces of stirrup from each other
Twist the strips so that sticky side faces bandage and stick to the conforming layer (Fig. 19.5j)  19.5j
Wrap protective layer over entire bandage working from distal to proximal
Limb is bandaged correctly – and is neat and tidy with no bits of cotton wool sticking out (Fig. 19.5k)  19.5k
Ensure bandage is not too loose. Check by flicking bandage – supposed to sound like flicking a watermelon!

- Repeat layers of cotton wool at least two more times.
- Incorporate traction tapes into layers to prevent slipping.
- Cover bandage with Elastoplast™ or cohesive outer layer.
- Leave distal end of middle two toes free in order to check circulation.

Ehmer sling

An Ehmer sling is used to prevent dislocation of the hip joint.

- Place animal in lateral recumbency with affected leg uppermost.
- Apply padding to metatarsus and stifle.
- Flex the leg and rotate the foot inwards.
- Apply conforming bandage to the metatarsus bringing it medial to the stifle.
- Continue over thigh, behind the hock and back to the metatarsus in a figure-of-eight pattern.
- The whole bandage may then be strapped to the body using cohesive tape.

Velpeau sling

This is used to support and immobilise the shoulder joint.

- Apply layer of padding material to the foreleg.
- Hold the paw in flexion and apply the bandage from the paw, over the back of the animal behind the other foreleg and back to the flexed paw.
- Continue in this fashion to stabilise the limb.
- Cover with adhesive or cohesive bandage.

Spica splints

Spica splints are used to immobilise the forelimb and shoulder. The bandage extends from the end of the foot to over the shoulder and around the body (Fig. 19.6).

Fig. 19.6 Spica splint.

WOUND BREAKDOWN AND DELAYED HEALING

It is absolutely essential that the VN is able to recognise the clinical signs of wound breakdown and delayed healing. The VN must be confident in examining wounds and reporting findings to the attending veterinary surgeon.

Clinical signs of wound breakdown include:

- erythema;
- inflammation;

- discharge;
- separation of wound edges;
- interference with the wound by the patient;
- systemic illness, e.g. pyrexia;
- change in wound colour (pale indicates reduced blood supply, dark/black indicates necrotic tissue);
- malodour.

Factors that affect the speed of wound healing include:

- the general physiological condition of the animal;
- the nutritional status of the animal;
- the blood supply to the wound area;
- presence of foreign material;
- presence of infection;
- the site of the injury;
- tissue mobility.

Chapter 20

Dentistry

THE VETERINARY NURSE'S ROLE

Extraction of teeth is an act of veterinary surgery and legally should only be performed by a veterinary surgeon. There are many hazards associated with a problem extraction, including gum damage, broken tooth roots and fractured jaws. Extraction by the VN is only acceptable if the tooth can be pulled out with the fingers.

Periodontal disease affects more than 85% of dogs and cats over 3 years of age. Although dentistry is a relatively new phenomenon to veterinary practice, many practices spend 25% or more of each day's operating time carrying out dental procedures. This has opened up a new role for the VN who plays a part in both the education of the client regarding tooth care and oral hygiene, and working with the patient and maintaining dental equipment. With the changes to the Veterinary Surgeons Act, VNs have more flexibility within dentistry and many practices have a specific dental nurse, who carries out mouth examinations and descaling. It is therefore essential that the VN is fully familiar with the terminology, anatomy and physiology associated with the oral cavity and the nursing considerations of the patient undergoing dental treatment.

COMMON TERMS AND CONDITIONS

Periodontal disease: an inflammatory response caused by residual food, bacteria and calcium deposits (tartar) that collect in the spaces between the gum and lower part of the tooth crown. If left untreated, infection can spread to the bone in which the teeth are rooted. The bone then resorbs and the teeth are slowly detached from their supporting tissues.

Halitosis: bad breath, which can be due to dental disease or uraemia.

Dental calculus (tartar): a combination of calcium phosphate and carbonate with organic matter, deposits in a hard covering on to the tooth surface.

Gingivitis: inflammation of the gums. There are numerous causes but it can lead to periodontitis.

Caries: erosion of tooth enamel because of bacteria from trapped food. Oxytetracyline given to pregnant bitches will affect the puppies' enamel causing pitting and yellowing of teeth and distemper also causes pitting and discoloration of the enamel in young dogs.

Fractured teeth: this can be caused by stone chewing. The teeth may need removing or repairing if painful.

Malar (cheekbone) abscess: infection of the tooth root of the upper carnassial tooth.

Malocclusion: this refers to malposition of the teeth resulting in the faulty meeting of the teeth or jaws. Malocclusion of the incisors is a common defect in all species and is inherited in many dog breeds, such as the brachycephalic breeds. With malocclusions in dogs and cats, abnormal striking of the teeth results in abnormal enamel wear. In mammals with open-rooted teeth (e.g. rodents) malocclusion is a problem because their incisors do not meet to cause the continual grinding down of their teeth that is required to prevent overgrowth and consequently dysphagia. In such cases, the incisors must be kept at an appropriate length by using dental burrs or files; clipping can shatter the tooth and produce sharp points which can damage surrounding tissue.

TREATMENT OF PERIODONTAL DISEASE

The following equipment should be prepared and set up prior to any dental procedure:

- ultrasonic dental machine containing a 0.2% chlorhexidine wash/coolant solution;
- dental polisher;
- dental forceps, hand-held scalers and curettes;
- mouth gag of appropriate size for the animal;
- pharyngeal packs;
- small pieces of damp cotton wool for cleaning debris;
- dental polish;
- fluoride solution;
- towels and grids to allow drainage of excess fluid from the mouth.

Scaling

The current standard treatment of periodontal disease is to scale the teeth to remove supragingival plaque and calculus that attaches to the root surface. If large deposits of calculus are present, the worst of it can be removed using dental forceps or rongeurs to snap off as much as possible. If this technique is carried out incorrectly, damage to the crown, enamel or gum tissue can occur. Once the worst of it is removed, an ultrasonic or sonic tooth descaler can be used. These tools work by vibrating a scaling tip at a high frequency over the tooth surface. This provides a fast method of scale removal but a certain amount of heat is generated which can cause tooth damage by causing pulp hyperthermia. When using these machines it is important to adhere to the following guidelines.

- Never spend more than 5–10 seconds per tooth. If necessary, go on to the next tooth and return to the original tooth in 10 seconds.
- Never press hard with the tip of the scaler.

- Never push the tip under the gumline (unless it is specially designed for this purpose).
- Hold the probe in a pen like grip and apply the tip side-on to the tooth (see manufacturer's instructions).

The desired effect is to achieve no visible debris on the tooth and the tooth surface is felt to be smooth and devoid of deposits and overhangs.

Hand scalers may be used instead of or in conjunction with powered equipment if desired. When properly performed, some dentists say that hand scaling is preferable to ultrasonic scaling because it causes less damage to the tooth structure. However, special training on hand scaling is required. Hand scalers are held using a pen grip and should be sharpened after each use. They should never be used under the gum line.

After scaling has been completed on all aspects of the tooth surface, a periodontal probe is used to feel for subgingival calculus and pockets greater than 3–5 mm in depth. Pocket depths are noted on the dental record.

Any remaining tartar from the root surface should be removed using subgingival curettes. If it is left, remaining tartar forms an inflammatory focus and the disease will continue to deepen the periodontal pockets.

The subgingival curettes have half-moon shaped tips which should be pushed into the bottom of the pocket and pulled upwards to remove the subgingival tartar. Overlapping strokes round the tooth in one direction and then again in the other should be performed to remove all of the calculus.

Polishing

Polishing is an essential part of the cleaning and scaling process because it smoothes out the tiny pits and fissures on the tooth enamel which are created by the scaling process.

Most dental machines have a separate handpiece to which a disposable rubber cup is attached. A prophy paste is used in combination with a short (no more than 5–7 seconds on each tooth) light polishing of each tooth. The rubber cups should be allowed to flare out on the tooth surface so that both the supragingival and subgingival crown enamel is polished. The cup should be thrown away after each patient.

Rinsing

After polishing, any remnants of prophy paste and other debris (e.g. calculus) are rinsed from the mouth and subgingival area. A 0.2% solution of chlorhexidine in a syringe with a blunted 21 g needle should be used to flush the gingival pockets around each tooth.

Fluoride treatment

It has been recommended that a final application of fluoride (FluroFoam™ mousse) should be sprayed into the mouth and left for 2 minutes before being washed off. This helps to harden the enamel and desensitise the exposed dentine.

Fig. 20.1 Dental sponges.

DENTAL CHART

A dental chart should be used to make notes about the amount of dental plaque, gingivitis and calculus, together with information about pocket depth, missing teeth, extracted teeth, malocclusions and gingival recession. These charts are available from most suppliers of veterinary dental equipment. They show a lateral and occlusal view of the mouth and each quarter of the mouth is given a number from 1 to 4: 1 = right upper, 2 = left upper, 3 = left lower and 4 = right lower. The numbering of the teeth starts from the midline, with the central incisor as 1.

ANAESTHETIC CONSIDERATIONS

A cuffed endotracheal tube must always be used and the cuff must be inflated. The pharynx should be packed with swabs: special foam sponges with string attachments are available – Metropacks™ dental sponges (Fig. 20.1). The head should be kept lower than the neck during the procedure if possible to allow all fluids to drain out of the mouth. The head must be kept lower during the recovery period, until full consciousness is regained. Remember to remove pharyngeal pack at the end of the procedure.

PERSONAL HYGIENE AND SAFETY

The VN should wear a protective mask, disposable gloves and safety goggles during dental procedures to provide protection from contact with the aerosol

droplets created by the ultrasonic cleaner. These droplets contain high numbers of bacteria.

EQUIPMENT MAINTENANCE

Hand instruments should be carefully washed in a proprietary instrument cleaner immediately after use and then dried thoroughly. Scalers and subgingival curettes should be sharpened with an Arkansas stone and oil before being autoclaved and stored in a perforated metal dental instrument tray. The powered equipment requires cleaning after use. The unit, cables and handpieces should be wiped down with a cleaning fluid and disinfectant. A couple of drops of the manufacturer's oil or lubrication should be placed into the inlet port of the handpiece. The turbine is removed from the high-speed handpiece and sprayed with an aerosol lubricant into the opening of the turbine. The turbine is then replaced and pressure is applied to the footpad to ensure the oil penetrates the workings of the machine. The pressure is released from the water tank and it is emptied and allowed to dry thoroughly.

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SECTION 5

ANAESTHESIA

INTRODUCTION

A good understanding of anaesthesia in different species is a very important part of veterinary nurse training. Although it is the responsibility of the veterinary surgeon to choose appropriate drugs and to induce anaesthesia, monitoring the patient and ensuring the animal is kept asleep during the procedure is often the role of the VN. Current changes towards the regulation of nurses may lead to a greater responsibility in the anaesthesia of small animals.

Chapter 21

Introduction – Evaluation of the Patient

A medical history should be obtained, with results from previous laboratory analysis and current bodyweight. All details must be recorded as a basis for comparison during and after surgery so that subsequent abnormalities can be detected quickly. All patients should be thoroughly evaluated prior to anaesthesia to assess not only the system affected by the primary disease but also the animal's general health (Table 21.1). The VN must be confident in performing a general clinical examination (see Chapter 3). Many patients, particularly those undergoing routine, elective surgery, will be in good health and may be anaesthetised with minimal risks using standard anaesthetic protocols. Some patients, however, will be classified as medium to high risk cases; they will need to be stabilised and an appropriate anaesthetic regime must be designed for them. A note should be made of previous and current medication (some medications may affect anaesthetic protocols). It may be useful in some cases to determine whether the animal has had a blood transfusion in the past, as any subsequent transfusion will require cross-matching.

LABORATORY TESTS

Animals undergoing elective surgery, class 1 and 2 patients, can usually be safely anaesthetised using routine techniques, however, laboratory screening tests (packed cell volume, total serum protein and blood urea nitrogen) may be useful to diagnose an animal with subclinical disease. Older animals, those undergoing long surgical procedures, trauma patients and animals with abnormal organ function will benefit from a general biochemical and haematological profile prior to surgery to minimise complications and to help choose an appropriate anaesthetic protocol. Other tests and evaluation procedures, such as radiography to assess lung function and electrocardiography to evaluate the heart, may be indicated in the higher risk anaesthetic patients.

PATIENT STABILISATION

A certain number of cases requiring surgery will need some form of stabilisation prior to surgery, for example patients with hypovolaemic shock as a result of trauma (e.g. road traffic accident), septicaemia (e.g. pyometra) and endotoxaemia (e.g. gastric dilatation).

Table 21.1 Categorising an animal's physical status (adapted from American Society of Anesthesiologists).

Class	Condition of animal
Class 1 – minimal risk	Normal healthy animals with no underlying disease, e.g. ovariohysterectomy, castration, hip dysplasia radiography
Class 2 – slight risk	Animals with mild systemic disturbances with no clinical signs of disease because they are able to compensate, e.g. neonate or geriatric animals, obese animals, animals with fractures but no shock, mild diabetes
Class 3 – moderate risk	Animals showing mild clinical signs associated with moderate systemic disease, e.g. anaemic, anorexic, moderately dehydrated patients, low-grade kidney disease, low-grade heart disease, pyrexia
Class 4 – high risk	Animals with pre-existing systemic disease or severe system disturbances, e.g. severe dehydration, shock, anaemia, uraemia, toxaemia, pyrexia, uncompensated heart disease
Class 5 – grave risk	Animals with a life-threatening system disease, e.g. moribund patients, advanced heart, kidney, liver, lung or endocrine disease, profound shock, severe trauma

Fluid therapy

Fluid therapy is indicated in dehydration, trauma, shock or metabolic disease. By combining the information gained from an accurate history with evidence of any clinical signs associated with dehydration, the need for fluid therapy can be assessed.

Disturbances in the body's electrolytes and acid–base balance can produce a wide range of clinical signs. Large changes in blood pH may result in the animal becoming depressed and can be fatal. The aim of fluid therapy is to replace deficits and rectify the electrolyte and acid–base imbalance that may have occurred. The fluid therapy protocol can be planned to take any changes into consideration, but without the necessary analysing equipment an estimate may only be made using information gained from history, clinical examination and knowledge of disease processes.

WITHDRAWING FOOD AND WATER

Dogs and cats

It is usually recommended that food is withheld for 8–12 hours prior to induction of anaesthesia. Studies have demonstrated a wide range of gastric emptying times; the range may be from 6 hours up to 12 hours. Prolonged starvation is not appropriate in young animals or older animals with concurrent disease because these animals will have depleted liver glycogen reserves, which leave the animal less able to withstand the stress of anaesthesia and surgery. Water,

in the vast majority of cases, should only be withheld for 2 hours prior to anaesthesia and no longer to prevent the risk of dehydration and hypovolaemia.

Rabbits

Rabbits do not require fasting prior to anaesthesia as they have a tight cardiac sphincter which prevents vomiting. Rabbits often suffer from ileus post anaesthesia and therefore feeding up until about 30 minutes prior to anaesthesia should be allowed to try to prevent gut stasis. It is advised that fluids should be given prior to anaesthesia to prevent dehydration. Rabbits require a higher maintenance rate than dogs and cats and so rates of 75–100 ml/kg/24hr may be given intravenously using the marginal ear vein or cephalic vein. Bolus doses of 30–50 ml subcutaneously may be given prior to anaesthesia to assist maintenance of hydration in routine cases.

Guinea pigs

Guinea pigs also do not require extended periods of fasting prior to anaesthesia; however starvation of 1–2 hours ensures the mouth is clear of food as food is often held at the back of the pharynx and may interfere with intubation. Nasal and oral secretions often become more viscous during anaesthesia and therefore frequent aspiration may be necessary.

Rats and mice and other small rodents

Small rodents do not vomit and therefore pre-anaesthesia fasting is not necessary. Fasting these species can lead to hypoglycaemia due to their high metabolic rate.

Ferrets

Ferrets readily vomit on induction of anaesthesia and therefore food should be withdrawn prior to anaesthesia. It is recommended that food should be withdrawn up to 6 hours, although, again due to a high metabolic rate, 2–4 hours may be more realistic to prevent post-anaesthesia hypoglycaemia.

Reptiles

Snakes require up to 2 days fasting prior to anaesthesia to ensure regurgitation is prevented. Absence of food in the gut also reduces pressure on the lungs and/or heart.

Chelonian species rarely regurgitate and do not require pre-anaesthetic fasting. Reptiles' low metabolic rate often poses a problem with the anaesthesia drugs used, as recovery can take up to 24 hours.

Birds

Avian patients have a high metabolic rate and therefore extended pre-anaesthetic fasting may lead to severe hypoglycaemia which can often be fatal

in the very small breeds. It is important, however that the crop is empty prior to anaesthesia, so 1–3 hours' fasting is recommended (the longer time being for birds >250–300 g).

PREMEDICATION

Pre-anaesthetic medication is frequently administered prior to induction of general anaesthesia for a number of reasons:

- to calm and control the patient;
- to provide analgesia throughout surgery;
- to reduce the amount of anaesthetic induction and maintenance agent required;
- to produce a smooth induction of anaesthesia;
- to reduce some of the unwanted side-effects of some of anaesthetic agents;
- to ensure a smoother recovery from anaesthesia.

A combination of sedatives/tranquillisers, analgesics and anticholinergic agents is most frequently used. There may well be a standard practice pre-medication protocol, which is suitable for most surgical cases. However, the choice of drugs will depend on the patient's physiological condition and the type of surgery to be performed.

The VN must be confident in calculating dose rates, be familiar with administration routes and be aware of common side-effects of the drugs used. Factors determining the choice of pre-anaesthetic drugs include:

- facilities and drugs available;
- experience of the veterinary surgeon and nurse;
- postoperative facilities;
- species variation (e.g. certain drugs are contraindicated in some species, and drug doses differ in different species according to their metabolism);
- general condition of the animal and other diseases;
- age;
- temperament;
- breed peculiarities (e.g. Boxers and acepromazine);
- proposed surgical procedure:
  - site;
  - need for muscle relaxation;
  - type (Caesarean, thoracotomy);
  - duration.

INTRAVENOUS CATHETERISATION

If possible, an intravenous (IV) catheter should be placed prior to surgery. This may be easier to carry out after the animal has been premedicated, although the hypotensive effect of many drugs may make catheter placement

more difficult. The presence of a catheter line facilitates a smoother induction if an intravenous agent is to be used and greatly reduces the risk of accidental perivascular injection. Intravenous fluid therapy can also be administered via this route. Should intravenous fluid therapy be deemed necessary during surgery, a pre-placed catheter ensures quick and easy administration. Catheterisation also provides a swift administration route for drugs in the event of an anaesthetic emergency. See Chapter 40 for technique.

POST-ANAESTHESIA RECOVERY

Post-anaesthesia care and observation of the patient are essential parts of the whole surgical procedure and should never be underestimated. The VN must be confident about which parameters to monitor and be aware of the common complications associated with each type of surgical procedure and each individual patient. Routinely, a temperature, pulse and respiration rate must be obtained and recorded every 15 minutes until a return to normal is seen. The patient should never be left unattended whilst the endotracheal tube is in place.

Hypothermia

Body temperature is regulated by the hypothalamus. The responses of the central nervous system are depressed in the anaesthetised patient and therefore the core body temperature drops. Additional factors include the cold anaesthetic gases administered via a non-rebreathing circuit. Surgical cases are also prone to heat loss due to the combined effects of the presurgical clip and scrub, and cold operating rooms and tables. In addition, reduction in muscular activity during anaesthesia and an anaesthetic-induced decrease in the basal metabolic rate also induce hypothermia. It is also the most common cause of bradycardia following surgery and a delay in recovery time. Hypothermia slows down the metabolic rate and therefore the metabolism of anaesthetic drugs. Methods for rewarming or preventing hypothermia are discussed in Chapter 5.

Postoperative pain

Prevention of pain is far better than trying to achieve adequate pain relief once the animal perceives pain. Knowledge of the type of surgery or procedure is important to ensure the premedication includes appropriate analgesics. Opioid analgesics provide the most effective means of pain control in small animals. In cases where there is impaired respiratory function, lower doses of opioids or alternative agents should be used. Partial opioid agonists such as buprenorphine can be slow to act but have a reasonably long effect; there is evidence that efficacy decreases with increased dose (the bell-shaped dose–response curve). Non-steroidal anti-inflammatory drugs (NSAIDs) only relieve pain of low to moderate intensity. When NSAIDs are used as analgesics, care must be taken especially in patients with renal, hepatic or cardiac dysfunction.

Chapter 22

Inducing General Anaesthesia

The VN is required to assist with all areas of anaesthesia from pre-anaesthetic checks and premedication, as discussed in Chapter 21, to induction, monitoring and maintenance and finally recovery of the animal.

PREPARING EQUIPMENT

It is essential all equipment is prepared and checked prior to use.

- Anaesthetic machine: oxygen supply? Flowmeter works? Vaporiser full? Correct inhalational agent?
- Breathing system: correct system selected? Checked for leaks? Valve open? Calculated flow rate?
- Other equipment.
  - Selection of endotracheal tubes (cuffs inflated and checked for leaks, check tube not blocked – especially small cat tubes).
  - Laryngoscope and blade?
  - Induction agent – calculated dose?
  - Heparinised saline flush.
  - Bandage to tie the tube in?
  - Cuff inflator?
- Emergency drugs and equipment to aid difficult intubation?

INDUCTION OF ANAESTHESIA

The premedicant drugs should have made the animal calm and sedated prior to induction. The intravenous catheter should be flushed prior to injecting the anaesthetic agent to ensure it is patent. The calculated dose is then administered either incrementally or as a bolus depending on the drug used and the effect of premedication. The room should be quiet at this point to ensure a smooth induction. The jaw reflex should be absent to allow intubation. Cats are susceptible to laryngeal spasm, so local anaesthetic is sprayed on the vocal chords to desensitise them prior to intubation. The endotracheal tube is placed and the breathing system attached and oxygen administered. The rebreathing bag may be squeezed to check for leakage around the tube, and the cuff is inflated accordingly. At this stage the VN checks the pulse and ensures the animal is breathing; some drugs cause ‘induction apnoea’ or the animal may be too deep and not breathing.

ENDOTRACHEAL INTUBATION

The endotracheal tube provides a connecting link between the patient's airway and the anaesthetic system. It also maintains a patent airway and can be used to manually ventilate the patient if necessary. VNs must be confident in carrying out intubation so that it may be performed efficiently in the event of an emergency or if the tube becomes dislodged during surgery and the veterinary surgeon is unable to de-scrub.

Tube selection

- Sizes range from 2.0 mm to 16 mm (internal diameter) for small animals.
- The tube should extend from the nose to a point level with the spine of the scapula.
- Tubes should be cut to the correct length before use. If the tube is too long, the dead space will be increased.
- Uncuffed tubes are recommended in cats as the trachea can be damaged easily by overinflated cuffs. It also means a tube with a larger internal diameter may be placed, decreasing resistence. Cuffed tubes may be necessary to ensure a good seal if the cat is to be ventilated during the procedure.
- Reinforced tubes help prevent kinking when the neck is flexed, e.g. during cisternal puncture or positioning for certain surgical procedures.
- Inflate the cuffs of the tubes selected prior to induction of anaesthesia to ensure the cuff is functional and there are no leaks.

Intubation technique

Assistant restraining the animal

(1) Restrain animal in sternal or lateral recumbency (depending on operator's preference).
(2) Extend the animal's neck.
(3) Open the animal's mouth with one hand, usually left hand if the animal is in left lateral recumbency. Avoid putting fingers in the animal's mouth – place thumb on left gum and index and middle finger on the other gum. An alternative technique is to pass a tape strip in the mouth, behind the canines to hold the upper jaw open – this is especially useful in brachycephalic dogs and reduces the risk of the animal accidentally biting the assistant or operator (Fig. 22.1).

Operator

(1) Always ensure there is adequate anaesthetic depth to ensure muscle relaxation of the upper airway.
(2) Select an appropriate size of endotracheal tube. Using a tube that is too large may cause laryngeal or tracheal trauma. A tube that is too small will enable the animal to breathe room air around the tube or allow aspiration of oral secretions.
(3) Check the length of the tube before use – from the nostrils to the manubrium. Cut the tube down if necessary.

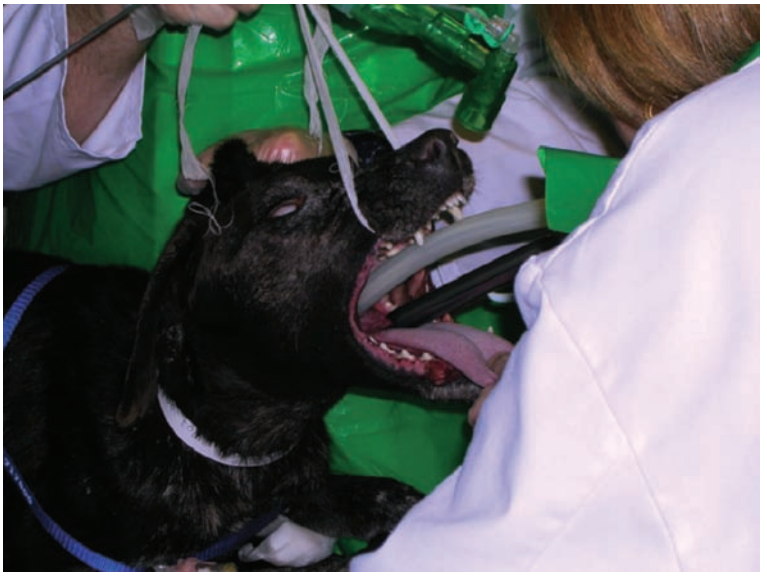

Fig. 22.1 Using a tape to hold the jaw open for intubation.

(4) Check the function of the cuff before intubating the animal.
(5) Lubricate the tracheal end of the tube with water or water-soluble gel.
(6) Pull the animal's tongue forward out of the mouth with one hand, using one finger of the same hand to pull the mandible downwards to open the mouth as much as possible. Take care not to lacerate the ventral aspect of the tongue on the lower incisor teeth. This may be prevented by placing one finger behind the lower canines and under the tongue.
(7) If necessary position light source to view larynx. Place the tip of the laryngoscope blade at the base of the tongue and press the tip of the blade ventrally to expose the glottis.
(8) Apply local anaesthetic if necessary. Cats are particularly prone to laryngospasm and therefore require local anaesthesia of the vocal folds.
(9) Depress the epiglottis with the tip of the endotracheal tube if necessary.
(10) Hold the tube so that the bevel is in the vertical position in line with the vocal folds and cavity.
(11) Pass the end of the endotracheal tube through the glottis into the trachea.
(12) Once the bevel is positioned through the vocal folds into the larynx, turn the tube so that the curve of the tube matches the curve of the trachea.
(13) Continue to advance the tube carefully until the end is level with the nose.
(14) The assistant places the animal's head gently on to the table.
(15) Check to ensure correct placement of tube. This may be performed by partially closing the adjustable pressure limiting (APL) valve on the circuit and squeezing the bag whilst the assistant listens for air escaping around the tube and inflates the cuff accordingly.
(16) Tie white open-wove tape strip securely to the endotracheal tube.

(17) Secure the tube in position by tying behind the ears or upper or lower jaw depending on procedure to be performed or size of animal.
(18) The trachea may relax slightly once the animal is more deeply anaesthetised, so further inflation of the cuff may be necessary, taking care not to overinflate as this may reduce the internal diameter of the tube and cause tracheal damage.

Notes:

- Forced intubation may cause damage to the larynx resulting in oedema, laryngeal spasm, vagal stimulation and cardiac arrhythmias.
- Local anaesthetic agent may be used in cats to reduce laryngospasm.
- NEVER press on the thorax of the anaesthetised patient to check that the endotracheal tube is in place. This can inadvertently cause gastric reflux and result in oesophagitis or, worse, aspiration if the tube is not a good fit.

Difficult intubations

Patients with airway disease or potentially difficult airways (brachycephalic breeds and fractured or dislocated jaws) require careful planning of their anaesthetic protocol. Drugs are chosen to ensure a rapid recovery and patent airway. It may be necessary to prepare for an emergency tracheotomy if oral intubation cannot be performed. Any cases which may prove to be difficult to intubate should be pre-oxygenated for at least 10–15 minutes prior to inducing anaesthesia. This ensures the patient is adequately oxygenated (100%) during intubation. This is also important in small mammals where intubation may not be straightforward.

In cases where the airway is narrowed or difficult to visualise and the endotracheal tube does not advance easily, a guide wire maybe used. This may be a narrow bougee or urinary catheter which is placed in the trachea and then the endotracheal tube is threaded over the guide wire, carefully advancing it into the trachea. It may require a much smaller diameter endotracheal tube to be placed than presumed for the size of animal.

EXTUBATION

(1) Leave the tube in place until the swallowing reflex returns. In the cat, the tube should be removed just prior to this to prevent laryngeal trauma.
(2) Ensure the cuff is deflated before removal. Where there are increased secretions or blood present, the cuff may be left slightly inflated to reduce the risk of aspiration of mucus or blood.
(3) Pull the tongue forward and gently extend the neck.
(4) Observe the patient to ensure the airway remains clear.
(5) After use, the tubes should be washed and disinfected. Oral secretions should be removed from the inner lumen using a bottlebrush and the outside of the tube gently wiped. The whole tube should be rinsed with water and the cuff tested before leaving it to soak for an appropriate time in disinfectant solution (e.g. Virkon™).
(6) The tubes can be sterilised using ethylene oxide (Anprolene™) and the latex tubes can be autoclaved, although this quickly perishes the rubber.

Chapter 23

Monitoring Anaesthesia

Monitoring general anaesthesia is a full-time responsibility and should not be combined with other duties or tasks. It is important to recognise the different signs associated with changing levels of unconsciousness in the patient, and be able to take the appropriate actions required. When an animal is anaesthetised, it is taken from a state of consciousness to unconsciousness, stopping at surgical anaesthesia before returning to consciousness again. When the VN monitors an animal during this time, he or she needs to be able to ensure that the animal remains stable at the surgical anaesthetic level. Overdose and failure to recognise signs of deepening unconsciousness can lead to death. It is important to recognise the different signs associated with changing levels of unconsciousness in the patient, and be able to take the appropriate actions required. Early observation and appropriate intervention will help to ensure that minor problems do not turn into major anaesthetic emergencies.

COMPLETING THE ANAESTHETIC RECORD

The anaesthetic record is a legal document and should be completed correctly and with full detail (Fig. 23.1). It may need to be referred to in cases of litigation.

The patient should be checked frequently (at least every five minutes) and the results recorded on an anaesthetic chart. This record encourages regular checking of the patient, allows trends to be identified and provides evidence in the event of litigation. All drugs that are given during the anaesthetic should be recorded (dose in milligrams, time given, route etc.). Any other event that may have occurred (extubation during radiography, moved to another room etc.) should also be recorded. The patient should be watched particularly closely during induction and recovery from anaesthesia.

MONITORING ANAESTHESIA IN DOGS AND CATS

Quick guide to monitoring an anaesthetised animal

- Watch and monitor the animal at all times during the anaesthetic. Do not leave the animal at any time.
- Place an oesophageal stethoscope for every anaesthetic.

ANAESTHETIC RECORD

Owner:
Date:

Animal name:

Breed:

Sex:

Age:

Weight:

PRE-OP CHECK

Temp. **Pulse** **Resp.** **MM/CRT:**

Premed:

Drug: Dose:

Drug: Dose:

Induction agent:

Drug: Dose:

ET tube size: Circuit:

TIME																		
Volatile agent %																		
Oxygen (l/min)																		
sPO_2																		
$ETCO_2$																		
160																		
140																		
120																		
100																		
80																		
60																		
40																		
20																		
0																		

Fig. 23.1 Example of an anaesthetic record.

Table 23.1 Normal values of vital parameters for patients.

Parameter	Dog	Cat
Heart rate	80–140 beats/min	110–140 beats/min
Respiration rate	10–30 breaths/min	20–40 breaths/min
Temperature	36–38°C	36–38°C
Capillary refill time	<2 secs	<2 secs
Oxygen saturation	>97%	>97%
Diastolic pressure	60–100 mmHg	60–100 mmHg
Systolic pressure	110–160 mmHg	110–160 mmHg

- Obtain the following vital signs at the start and then at least every 5 minutes throughout the procedure (see Table 23.1):
  - heart rate;
  - respiration rate;
  - capillary refill time;
  - pulse quality.
- Obtain a core temperature at the start and then at the end of the procedure.
- Write down the findings on an anaesthetic chart.
- Check the position of the eye (should be rotated ventromedially but may vary).
- Check the palpebral reflex (should be absent during procedure).

The following should be checked to assess depth of anaesthesia:

- respiratory system;
- cardiovascular system;
- reflexes;
- mucous membranes;
- temperature.

Respiratory system

It is important to ensure that there is a patent airway during anaesthesia, and that adequate ventilation is being achieved. The VN should check the respiration movements by looking at the bag on the anaesthetic machine. This enables the VN to monitor the respiratory rate without disrupting or touching the surgical drapes and, more importantly, indicates that the patient is still connected to the anaesthetic machine and that there is not an obstruction anywhere.

Respiratory rate

The respiratory rate under anaesthesia should be similar to the resting rate in the conscious animal, although it is often reduced during general

anaesthesia especially following induction with barbiturates. An increase in rate usually indicates that the patient is becoming light and a decreased rate usually indicates an increase in level of unconsciousness. It is important to remember, however, that an increased rate may also be seen in a hypoxic animal because it is trying to obtain more oxygen.

Respiratory depth

The depth of respiration is important, and the VN should be able to see good movement of the chest wall and the bag on the anaesthetic circuit, and ensure that the patient is receiving its required tidal volume (10–15 ml/kg) by checking the minute flow rates. A Wright's respirometer can be used to determine the tidal volume.

Respiratory pattern

The pattern of ventilation during anaesthesia should be regular. As the level of unconsciousness increases, respiration becomes irregular and the patient may gasp until respiration ceases altogether. The respiration pattern usually becomes irregular and faster if the animal is insufficiently anaesthetised.

Cardiovascular system

Heart rate

It is important that cardiac output is maintained during anaesthesia to ensure adequate perfusion of tissues and vital organs (brain, heart, liver and kidneys). By listening to the heart sounds and feeling a pulse, the VN will be able to detect subtle changes early on and take appropriate steps to maintain cardiac output (e.g. lightening the volatile agent, initiating intravenous fluid therapy). Every anaesthetised animal should have an oesophageal stethoscope placed to facilitate constant heart rate and rhythm monitoring. An electrocardiogram (ECG) can indicate electrical activity and, by counting the complexes on the trace, the heart rate can be assessed. The ECG electrodes should be placed on the patient's limbs, on the forelimbs behind and slightly proximal to the elbow, on the hindlimbs cranial and proximal to the stifle. Good electrical contact should be ensured using either gel or surgical spirit. In the UK the following system is used:

- red lead – right fore;
- black lead – right hind;
- yellow lead – left fore;
- green lead – left hind;

It is important that these leads are positioned correctly to get a true reading.

Pulse

The VN must start palpating a pulse at the beginning of anaesthesia and continue regularly throughout the procedure so that subtle changes can be detected. Palpation of the pulse allows the heart rate and the pulse quality to

be measured. A strong and easily palpated pulse indicates good pressure and good cardiac output. In contrast, a weak, thready pulse indicates hypotension, hypovolaemia and increase in the level of unconsciousness. It is advisable to palpate a peripheral pulse rather than a central pulse. This is because peripheral pulses are more sensitive to changes in the circulation and therefore changes can be detected earlier.

Peripheral pulse sites include:

- sublingual (ventral midline surface of the tongue);
- dorsal pedal/metatarsal artery (caudomedial surface of hock);
- digital/brachial artery (palmar aspect of the carpus).

A central pulse site is the femoral artery (medial aspect of the proximal thigh).

Rhythm

The rhythm of the heart and pulse should be regular and constant. Dysrhythmias may indicate deep levels of unconsciousness and/or hypovolaemia. An ECG is useful to detect cardiac dysrhythmias and heart rate. It is, however, important that the VN is aware that it is possible for an ECG trace to be relatively normal even if the heart muscle is not contracting at all (electromechanical dissociation which is now referred to as pulseless electrical activity (PEA)). For this reason, the VN should not rely on ECG traces alone as a monitoring method.

Blood pressure

Arterial blood pressure is monitored by nerve endings in the arterial wall (baroreceptors). These monitor the blood pressure and make changes accordingly (vasoconstriction, vasodilation, increase in heart rate, etc.). During anaesthesia, these responses are depressed. Measurement of blood pressure gives a clear indication of the peripheral circulation and can indicate more accurately whether the patient is hypotensive or hypertensive. Maintenance of blood pressure is important so that vital organs are adequately perfused. A low blood pressure (mean <60 mmHg) may indicate hypotension. Many anaesthetic drugs cause a dose-dependent fall in blood pressure and this should be taken into consideration. Blood pressure can be monitored directly by an arterial cannula connected to a pressure transducer or indirectly by using an occlusive cuff and Doppler ultrasound detectors or an oscillometric machine. The Doppler device detects an ultrasound echo from the blood passing through the vessel.

The most common sites for blood pressure measurement in the dog and cat are:

- brachial artery (palmar surface of forepaw);
- metatarsal artery (caudomedial surface of hindpaw);
- saphenous artery (plantar surface of hindpaw proximal to foot pad);
- coccygeal artery (ventral surface, base of tail).

Measuring blood pressure using a doppler ultrasound probe.

(1) Clip the hair over the chosen artery.
(2) Ultrasound gel is applied to the probe which is then placed over the artery and taped in place.
(3) The pulse may be heard as a swooshing sound through the speaker or earphones.
(4) Select a blood pressure cuff of appropriate size (width of cuff should be 40–60% circumference of limb).
(5) Place the cuff just proximal to the probe and secure.
(6) The cuff is connected to an aneroid manometer and the bulb inflated until the artery is occluded and the pulse cannot be heard.
(7) The pressure in the cuff is slowly released until the first sounds of blood flow are detected. The pressure recorded at this time is the systolic pressure. As the cuff pressure is continued to be released the second sound is heard; the pressure recorded at this time is the diastolic pressure.

Mucous membranes

Observation of the colour of the mucous membranes is extremely useful to assess the condition of the patient's circulation. The mucous membranes should be pink, but, as with many other signs, it is important to know the colour that they were before the start of anaesthesia so that it is possible to make a comparison. Pale mucous membranes indicate hypotension/hypovolaemia due to a fall in peripheral perfusion. Blue (cyanotic) membranes indicate that the patient is being inadequately oxygenated. Remember, however, that the mucous membranes are slow to react to hypoxia and cyanosis is really only ever seen in severe cases of hypoxia.

Capillary refill time

Capillary refill time (CRT) should be less than 2 seconds. Increased CRT may indicate hypotension, hypovolaemia, toxaemia and/or haemorrhage.

Monitoring the nervous system – reflexes

As the level of unconsciousness deepens, so muscle tone is lost. Some of the following reflexes are useful as a way of assessing the level of unconsciousness, but they should always be used in conjunction with other observation methods.

Jaw tone: not very accurate or sensitive, it is soon lost after induction.
Pedal withdrawal reflex: not very accurate or sensitive, again it is soon lost after induction.
Palpebral reflex: this is the most useful of all of the reflexes to check. It can be done by gently tapping the medial canthus of the eye or by gently brushing the animal's eyelashes. Used in combination with other signs, it can be used to assess the level of unconsciousness. The reflex is present in conscious animals and it becomes less apparent during anaesthesia. For most surgical procedures it is necessary that the palpebral reflex is absent. A

return of the reflex indicates a lightening of the anaesthesia or an insufficient level of unconsciousness.

Corneal reflex: a corneal reflex is present until very deep anaesthesia (death!) and should never be used to assess the depth of anaesthesia because the cornea can easily be damaged.

Pupil diameter: the pupil diameter does increase as the patient becomes more deeply unconscious. However, it does also depend on some of the drugs used in premedication regimes (e.g. atropine dilates the pupil whilst opioid drugs constrict the pupil).

Eye position: the eye rotates down and medially during anaesthesia. If it returns to normal position during anaesthesia, it could indicate that the animal is becoming too light or too deep. Usually, if the animal is too light, the eye returns to normal position and a palpebral reflex is present. If the animal is too deep, the eye returns to its normal position, but there is no palpebral reflex and the pupil tends to dilate. This may also be accompanied by a drying out and wrinkled appearance of the corneal surface.

Temperature

It is important to obtain a core body temperature before anaesthesia and then regularly throughout and after the procedure. During anaesthesia, patients inevitably become hypothermic. This is due to a combination of factors, including an anaesthetic depression of the hypothalamus (decreases temperature regulation), cold anaesthetic gases, cold theatres, large clipped areas on the body, cold wet drapes and cold wet solutions used for cleaning the patient. It is essential to take steps to avoid hypothermia because it is not only uncomfortable and unpleasant for the recovering patient but it can also lead to prolonged recovery times, shock and death. This is particularly true in small dogs, cats and other small mammals.

MONITORING ANAESTHESIA IN OTHER SPECIES

Rabbits, ferrets, guinea pigs and other small rodents

Most important in the smaller mammals is maintenance of core body temperature and hydration. With high metabolic rates, it is equally important to ensure they recover quickly and return to normal eating and drinking as soon as possible.

Monitoring depth of anaesthesia is done by assessing the response of the animal to noxious stimuli. Other reflexes may also be useful in small mammals such as the pedal withdrawal reflex, although rabbits tend to retain this reflex until very deep levels of anaesthesia. Loss of reaction to the ear pinch in rabbits and guinea pigs, and the tail pinch in rats and mice usually indicates a surgical plane of anaesthesia.

Reptiles

Reptiles should be maintained at temperatures close to their optimal environmental temperature (around 22°C) and then slightly warmer (25–30°C) for

the recovery period. Maintaining hydration is also important to aid recovery and return to normality.

Ventilation may be necessary in chelonians as soon as anaesthesia is induced as respiratory movement is controlled by muscular contractions of the limb muscles as well as the respiratory muscles. Ventilation may be carried out by gently moving the legs in and out which changes the volume of the coelomic cavity.

The heart rate and respiratory rate may be monitored by an oesophageal stethoscope. It is also important to monitor the reptile's temperature and use methods to prevent the animal becoming too cold, which would slow metabolism and extend recovery times. An ECG and Doppler flow detector may be used, however a pulse oximeter is unreliable due to the thick skin (or scales).

Birds

Initially the respiration becomes irregular and shallow and slow voluntary movement is still seen. As a deeper plane of anaesthesia is reached the pedal reflex is still present but slow and the palpebral is lost. Respiration becomes deeper and more regular. Heart rate may be monitored easily with a stethoscope and ECG leads may be attached with pads or clips to fine needles in the skin.

Maintenance of core body temperature is very important in birds, especially the very small breeds, when their normal core temperature is around 40°C.

ANAESTHETIC MONITORING AIDS AND EQUIPMENT

As previously stated, all anaesthetised animals should have an oesophageal stethoscope placed so that the anaesthetist can listen to the heart and respiratory sounds frequently or, if necessary, constantly without disturbing the drapes and surgeon. Other monitoring aids such as electrocardiographs, pulse oximeters, respiratory monitors, blood pressure monitors and capnographs (carbon dioxide monitors) are extremely useful in the critical, high-risk patient. However, the most reliable and effective method of assessing levels of unconsciousness is by the anaesthetist physically observing and touching the patient.

Electrocardiograph: measures the electrical activity of the heart. Paper trace monitors are more useful for diagnosing heart rhythm abnormalities whereas the oscilloscope display is more useful for continual monitoring during anaesthesia. The VN should recognise a normal trace and alert the veterinary surgeon should any abnormalities arise, such as dysrhythmia or ectopic beats (an additional heartbeat originating from elsewhere on the heart).

Pulse oximeter: this monitors both the pulse rate and the arterial oxygen saturation of the haemoglobin in the blood. It is placed on the tongue, toe web or ear pinna. The normal value for an animal breathing room air is greater than 95%, but the anaesthetised patient on oxygen should be nearer 100%. Values less than 90% are seriously low, indicating inadequate oxygenation of the patient.

Arterial blood gas analysis: these machines can measure oxygen, carbon dioxide and also the pH of arterial blood. They are very useful for accurately monitoring respiratory function but are very expensive and so not routinely used in most veterinary practices.

Respiratory monitors: these have limited value as they indicate that the animal is breathing but give no indication of the tidal volume.

Capnograph: measures the end tidal carbon dioxide (CO_2) content. This approximates to that of arterial blood. A device is placed between the endotracheal tube and the anaesthetic circuit and volumes of up to 200 ml are sampled. The normal value for end tidal CO_2 is between 35 and 40 mmHg. Values above 50 mmHg indicate hypercapnia (high levels of CO_2 from hypoventilation, hypermetabolism).

ANAESTHETIC EMERGENCIES

Appropriate and consistent monitoring will enable the VN to be aware of potential problems occurring during anaesthesia. By alerting the veterinary surgeon immediately, anaesthetic-related fatalities may be avoided.

The quick guide to anaesthetic overdose or animal being too deep follows.

- Heart rate: reduced.
- Respiration rate: either reduced or panting/gasping.
- Pulse quality: rapid, weak, feeble pulse or none detectable.
- CRT: >2 seconds.
- Colour: pale or cyanotic.
- Eye position: up in the normal position.
- Palpebral reflex: absent.
- Pupil size: dilated (depends on drugs used).
- Temperature: reduced core temperature, animal feels cold to touch.
- Surgical wounds: reduced/no haemorrhage present.

Refer to Chapter 40 for resuscitation procedures.

Chapter 24

The Anaesthetic Machine

The anaesthetic machine has the following functions:

- to deliver inhalation anaesthetic gases;
- to deliver oxygen;
- to enable intermittent positive pressure ventilation.

All anaesthetic equipment should be checked regularly for faults. Faulty equipment should be labelled and replaced as soon as possible. It is important that the VN understands the function of the various parts of the anaesthetic machine to ensure they are working correctly.

GAS CYLINDERS

Inhalation gases are supplied under pressure, in metal cylinders of increasing sizes. The smaller of these cylinders (sizes AA to E) are mounted directly on to the anaesthetic machine via the pin–index system. The larger cylinders are used to provide piped gases. They are usually stored in a separate room within the building or in an outside lockup.

All gas cylinders are colour coded:

- oxygen: black with white top;
- nitrous oxide: blue;
- carbon dioxide: grey.

Some older machines have the facility to carry:

- cyclopropane: orange.

Different gases are supplied at specific pressures. The standard E size oxygen cylinders attached to anaesthetic machines are supplied at 134 bar pressure and contain 680 litres of oxygen. The pressure in an E size nitrous oxide cylinder is 44 bar which releases 1800 litres of nitrous oxide during use. At room temperature and pressure, nitrous oxide is a liquid. The liquid nitrous oxide then evaporates to form a gas as it is released. As the gas is used up, more liquid is vaporised. Nitrous oxide cylinders can be weighed to see how full they are.

The pin–index system

The cylinder valve face has two holes, which correspond to the pins on the machine. The holes for the nitrous oxide and oxygen cylinders are different to ensure the cylinders are not connected to the wrong port.

A BODOC valve or washer made from metal and neoprene ensures a firm seal between cylinder and machine. Oil or grease must not be used as a seal; the pressurised gases give off heat as they are released from the cylinder and may cause explosions if oil is used. Before attaching a full cylinder to the machine, briefly open and close the cylinder valve to clear any dirt from the port.

REDUCING VALVES/REGULATORS

Reducing valves or regulators are usually situated between the cylinder and the flow meter. As the pressure in the cylinder is high these regulators reduce the pressure in the cylinder to a lower working pressure (4 bar). They help to maintain a constant pressure to the machine. Non-return valves prevent empty cylinders still attached to the machine from refilling from fresh cylinders.

PRESSURE/CONTENTS GAUGES

Pressure or contents gauges may be combined with the reducing valve (compressed gas for surgical drills etc.). The oxygen pressure gauge indicates volume of gas remaining in the cylinder. The nitrous oxide pressure gauge will remain constant ('full') until the liquid evaporates when the pressure will fall rapidly. Never leave a an empty nitrous oxide cylinder on the machine overnight – the last remaining liquid vaporises causing the pressure gauge to read full!

EMERGENCY OXYGEN FLUSH

The emergency oxygen flush is also known as 'purge' or 'bypass' valve. Oxygen bypasses the vaporiser and exits through the common gas outlet at a rate of about 35l/min. It is operated by a spring loaded button so that it cannot be left on. If this flow is accidentally left on there is a risk of diluting the other anaesthetic gases. Beware of using this when the breathing system is attached to cat or small dog – it could blow their lungs!

FLOW METERS (ROTAMETER)

The flow meter controls the flow of gas indicating in 'litres per minute' (l/min) the quantity passing through the machine and to the patient. The level is indicated by a bobbin or ball which floats in a calibrated glass tube. Flow meters should be serviced regularly to check correct readings are given.

Bobbins and balls

Bobbin flowmeters are more accurate, but more expensive. The shape of the bobbin is designed to optimize gas flow, and has nothing to do with being a pointer. The flowrate is taken from the TOP of the BOBBIN, and the MIDDLE of the BALL-BEARING.

Inaccuracy in flowmeters

Inaccuracy may be due to:

- the tube not being vertical;
- back-pressure from, for example, a ventilator;
- static electricity causing the float to stick to the tube;
- dirt causing the float to stick to the tube – a small dot usually marks one side of the bobbin, so it can be seen to be rotating.

ANAESTHETIC VAPORISERS

The purpose of an anaesthetic vaporiser is to produce a controlled and predictable concentration of anaesthetic vapour in the carrier gas passing through the vaporiser. It is capable of delivering the volatile agent at various concentrations. The vaporisers must be used with the correct volatile anaesthetic and must not be interchanged. Vaporisers must be kept upright at all times to ensure the calibration is correct. Most vaporisers are of the plenum type, which consists of a vaporising chamber containing the liquid anaesthetic, and a bypass. Gas passing through the vaporising chamber volatilises the anaesthetic and is then mixed with the anaesthetic-free gas bypassing the chamber; the proportions of vapour-containing gas and bypass gas are controlled by a tap.

Factors affecting vaporiser output

Flow through the vaporising chamber

Vaporiser output is controlled by varying the proportion of gas passing through the vaporising chamber and bypass.

Efficiency of vaporisation

Vaporisers usually incorporate a system of wicks and channels in the chamber to improve efficiency of vaporisation and increase the output concentration of anaesthetic.

Temperature

As temperature increases, the output of the vaporiser will increase, unless some compensatory mechanism is used.

Time

Vaporisation causes the liquid anesthetic to cool since heat is lost because of the latent heat of vaporisation of the anaesthetic. Therefore, the output concentration will tend to fall over time. Some vaporisers are insulated to control this factor.

Gas flow rate

Changes in carrier gas flow rate may affect vaporiser output by:

- altering the proportion of the total gas flow that passes through the vaporising chamber;
- altering the efficiency of vaporisation; for example, at high flow rates, the gas leaving the vaporising chamber will tend to be less saturated (since the gas spends less time in the chamber), so the output of the vaporiser will tend to fall.

Older vaporisers are less accurate at lower flow rates.

Carrier gas composition

The composition of the carrier gas may affect vaporiser output by:

- changes in the viscosity and density of the gas mixture affecting the proportion of the total flow that passes through the vaporising chamber;
- nitrous oxide dissolving in the anaesthetic, thus altering the effective volume that passes through the vaporising chamber.

Ambient pressure

Saturated vapour pressure (SVP) is solely a function of temperature. Therefore, if ambient pressure is reduced, the (constant) SVP becomes a greater proportion of the total (reduced) pressure, and the ouput concentration (in volumes percentage) rises. For example, a halothane vaporizer calibrated at sea level and set to deliver 2% will deliver about 2.7% halothane if used in Denver, Colorado.

Filling vaporisers

Most newer vaporisers now have the additional health and safety feature of key filling. This prevents any of the volatile agents being spilt and evaporating into the environment. As well as being able fill the vaporiser correctly, you should also know how to empty it.

In-circuit vaporisers

- Stephen's or Komesaroff machine (Fig. 24.1).
- The patient's inspiratory efforts draw gas through the vaporiser.
- Deeper or more rapid breathing results in higher concentration.

Fig. 24.1 Stephen's in-circuit vaporiser.

When using an in-circuit vaporiser (i.e. a Stephen's or Komesaroff machine) remember the following points.

- If anaesthesia is too light, surgical stimulation will lead to an increase in ventilation and therefore a deepening of unconsciousness.
- If the vaporiser setting is too high, deepening anaesthesia depresses the ventilation and reduces vaporisation. This acts to some extent as a built-in safety factor.
- If the animal stops breathing then no fresh vapour enters the circuit.
- The smaller the fresh gas flow the greater the economy in the use of the volatile agent. (Ensure that oxygen requirements are met.)
- A simple, low-efficiency vaporiser is required (a Goldman vaporiser for halothane limits the concentration to less than 3%).
- A sudden increase in ventilation and, therefore of inspired concentration, may be dangerous.
- The fact that respired gases pass through the vaporiser introduces the problem of resistance to breathing.

WARNING DEVICES

Some machines are fitted with warning devices to indicate a lack of oxygen supply. This is obviously a huge advantage. Without doubt there have been many anaesthetic deaths or near-deaths through there being no alarm system and animals receiving either no oxygen supply or a lethal supply of nitrous oxide and halothane. When purchasing a new machine, always check that

there is an oxygen alarm device. All machines should be fitted with warning devices to indicate a lack of oxygen supply or too high pressure in the system.

Oxygen failure alarms

Most oxygen failure alarms are powered only by the oxygen pressure and do not depend on mains electricity or battery power. They are activated by a fall in oxygen pressure and emit a loud whistle that may only be reset by the return of the correct oxygen pressure. Until that time all the gases are vented to the atmosphere and away from the patient by a safety valve.

Bosun whistle

This is dependent on a flow of nitrous oxide. When the oxygen supply fails the nitrous will be cut off also and a whistle will sound. Newer machines have alarms that depend only on the falling oxygen supply.

Over-pressure alarm and pressure relief valves

High pressure, that may occur when breathing system valves are left closed, will sound an alarm.

If the anaesthetic machine is fitted with a pressure relief valve it will usually be located on the back bar distal to the vaporisers. The valves are designed to protect the machine and vaporisers against high pressures and not the patient. When the common gas outlet is occluded with a thumb, the pressure rises within the machine and will open a pressure relief valve, commonly at about 35 kPa. Testing for this alarm feature may be done by occluding the common outlet valve whilst depressing the oxygen flush button. Never try this in a machine without checking to see if a valve has been fitted.

CARBON DIOXIDE ABSORBERS

Rebreathing systems incorporate a carbon dioxide absorbent which consists of:

- sodium hydroxide (‘soda’);
- calcium hydroxide (‘lime’);
- silicates;
- pH indicator.

The colour change is:

pink to white
or
white to lilac.

The VN must ensure they know what colour the absorbent in the practice changes to. The colour change indicates that the absorbent is exhausted and

should be changed. If left overnight some absorbents change back to original colour. Soda-lime reacts with sevoflurane producing a harmful substance called compound A. Baralyme, containing 20% barium hydroxide and little alkali, may be used as an alternative to soda-lime. It is less dusty and the dust is less alkaline than that of soda-lime. Baralyme is also presented as small spheres instead of granules therefore creating less dust than soda-lime. The contents of a typical soda-lime canister will provide around 8 hours of use in small animals and it should then be changed whether or not it has changed colour.

Changing the absorbent

Soda-lime is an irritant alkaline substance. The dust MUST NOT be inhaled or allowed in contact with the eyes.

WEAR GLOVES AND A FACE MASK

when changing the soda-lime.

ANAESTHETIC SCAVENGING SYSTEMS

Importance of scavenging

COSHH Regulations 2002 require the prevention or control of exposure to waste gases.

Side-effects of waste gas pollution include:

- headaches;
- fatigue and nausea;
- spontaneous abortion;
- congenital abnormalities;
- reduced fertility;
- liver and kidney disease;
- neurological (nitrous oxide).

Methods of reducing pollution include:

- effective scavenging;
- maintenance of equipment;
- anaesthetic technique:
  - correct size endotracheal tube;
  - avoid poorly fitting face masks;
  - do not switch on vaporiser until patient is connected;
  - switch off vaporiser/nitrous before patient is disconnected;
  - empty rebreathing bag into scavenging system NOT into the room;
  - fill vaporisers at the end of the day;
  - avoid spillage of volatile agents;
  - flush the system with 100% oxygen before disconnection.

Concentrations of anaesthetic gases may be monitored and must not exceed the following limits:

- halothane 10 ppm;
- nitrous oxide 100 ppm;
- isoflurane 50 ppm.

(According to the Occupational Exposures Standards 1996.)

Methods of scavenging

Passive: a length of corrugated plastic tubing is connected to the scavenging valve of the anaesthetic system. The other end feeds through a hole in the wall to the outside. The tubing should not be longer than 2.4 m.

'Fluosorb': activated charcoal absorber – only absorbs halothane and other halogenated hydrocarbons NOT nitrous oxide. The container needs to be weighed regularly to check it is not 'full' (1400 g) and no longer working.

Active–passive: similar to passive system except the tubing discharges into a forced ventilation duct rather than directly outside.

Active: most effective method. A central vacuum removes the waste gases. Care must be taken when using this system with Ayre's T-pieces as the suction may be too great!

CHECKING AND SETTING UP THE ANAESTHETIC MACHINE

The anaesthetic machine must be thoroughly checked at the beginning of each day.

(1) Connect and switch on scavenging system.
(2) Check the vaporiser is full (should be filled the night before) and that the dial can be moved easily.
(3) Check the pressure of the cylinders and ensure a full spare is attached or available for use.
(4) Check that piped gas supply is switched on (where applicable).
(5) Check that the flow meters work.
(6) Check warning devices work.
(7) Check emergency oxygen flush works.

At the end of the day.

(1) Fill the vaporiser in a well ventilated room.
(2) Replace empty cylinders.
(3) Switch cylinders off and empty system of remaining gas.
(4) Clean and disinfect all surfaces.

Chapter 25

Anaesthetic Breathing Systems

Inhalation anaesthetic agents may be administered via anaesthetic breathing systems. There are various methods by which anaesthetic breathing systems are classified (American and UK writers have different definitions of 'closed' and 'semiclosed' systems and there are the Mapleson classifications). It is a good idea to be aware of the various terms, however. Hall and Clarke (2001) suggest that the most clinically useful method of categorising the different systems is based on the method by which carbon dioxide is removed from the circuit: *non-rebreathing systems* and *rebreathing systems*.

Non-rebreathing systems rely on a high fresh gas flow to vent expired gases out of the system into the atmosphere, whilst rebreathing systems pass the expired gases through a canister containing carbon dioxide absorbent which removes the carbon dioxide and allows the same gas to be rebreathed by the patient. Some of the systems are useful for providing manual ventilation (intermittent positive pressure ventilation – IPPV). See Table 25.1.

Whichever breathing system is selected, it must be able to deliver enough gas to prevent rebreathing and have as little resistance as possible to ensure adequate oxygenation of a range of different animals.

CALCULATING RESPIRATORY MINUTE VOLUME

$$\text{Minute volume} = \text{body weight (kg)} \times 10\text{–}15\ \text{ml (tidal volume)} \times \text{respiratory rate/minute}$$

Minute volume must then be multiplied by the anaesthetic system factor (see Table 25.1).

CHECKING THE SYSTEM FOR LEAKS

Whichever breathing system is selected, it must be tested for leaks before it may be used with a patient.

(1) Attach the breathing system to the common outlet on the anaesthetic machine.

Table 25.1 Which breathing system to use?

System	Patient size	Breathing system factor	IPPV?
Modified Ayres T-piece	<10 kg	2.5–3 × MV (Minute Volume)	Yes
Modified Bain	10–30 kg	2.5–3 × MV	Yes
Lack	10–40 kg	1–1.5 × MV	No
Magill	8–35 kg	1–1.5 × MV	No
Circle	10–100 kg	N/A	Yes
Mini Lack	<10 kg	200 ml/kg	No
Humphrey ADE	>5 kg	Depends on whether used as rebreathing or non-rebreathing	Yes

IPPV *may* be carried out with the LACK and MAGILL breathing systems as long as the flow rate is high enough.

(2) Attach the scavenging hose.
(3) Close the 'pop-off' or adjustable pressure limiting (APL) valve.
(4) Occlude the patient end of the breathing system.
(5) Depress the oxygen flush or turn the oxygen flow meter to 6–8 l/min until the rebreathing or reservoir bag is full.
(6) Turn off the flow meter.
(7) Gently squeeze the reservoir bag and listen for any leaks.
(8) Open the APL valve and squeeze the gas out through the scavenging system.

Note: when testing Ayres T-piece breathing systems, it maybe necessary to remove the APL valve first to check the bag as the lower pressure release on these valves makes it impossible to close the circuit fully.

NON-REBREATHING SYSTEMS

- Modified Ayre's T-piece (Mapleson D).
- Magill (Mapleson A).
- Lack (coaxial and parallel) (Mapleson A).
- Modified Bain (Mapleson D).

These systems work by providing a fresh gas flow, which is pooled into a reservoir from which the patient inhales the fresh gas and volatile agent combination. Expired gas from the patient is also passed into this area and then out into the atmosphere via a spill valve. For this reason, it is important that the fresh gas flow rates are high enough to help push out the exhaled carbon dioxide-rich gas and provide enough for the next inspiration. It is important that the patient's respiratory tidal and minute volumes are calculated and the gas settings fixed appropriately depending on the circuit used.

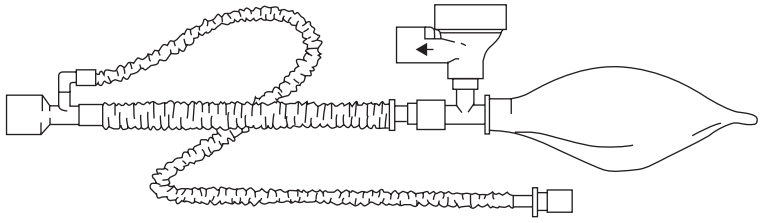

Fig. 25.1 T-Piece.

Ayre's T-piece (Mapleson 'E'); with Jackson-Rees modification (Mapleson 'F')

Originally there were no valves in this system, which means there was very little resistance to breathing, thereby making it the most suitable system for very small animals. As a general guide, this would be any animal weighing under 10 kg. The bag is called the Jackson-Rees modification and enables IPPV and can be used as a method of monitoring respiration. The most common T-piece used now has a valve situated between the closed end bag and the corrugate tube (Fig. 25.1). This is now very similar to the Mapleson D or modified Bain breathing system. The addition of the valve appears to provide very little additional resistance as long as flow rates are adequate.

To ensure exhaled gases are removed effectively enough to prevent rebreathing, the flow rate for this system should be 2.5–3 times the minute volume of the patient. Fresh gas passes along the narrow tubing from the anaesthetic machine towards the patient. Some of this gas passes into the corrugated tubing so that when the patient inhales it takes most of its gas supply from this area. Exhaled gases from the patient are also forced into this part of the system by the continuing fresh gas coming down tube. During the patient's expiratory pause, the exhaled gas is pushed out of the system via the bag, and a fresh supply of gas is collected ready for the patient's next inhalation. The main disadvantages of this system are the high gas flow rates required and the difficulty in scavenging waste anaesthetic gases.

Bain – modified Mapleson 'D'

Externally, the Bain looks almost identical to the coaxial Lack, however, this system works very differently and functions in a similar way to the T-piece. Fresh gas passes along the thin inner tube to the patient and expired gas passes through the outer sleeve to the bag and valve, which are placed on the machine end of the system (Fig. 25.2). Just like the T-piece, high gas flow rates are required to prevent significant rebreathing of expired gas. This is suitable for patients over 10 kg. The main disadvantage of this system is that very high gas flow rates are required, especially in heavier dogs, so effective scavenging is essential. Also, coaxial systems present an operational risk if the inner tube becomes detached because a very large dead space is created. This should be checked on a regular basis by occluding the patient end and passing 61/min

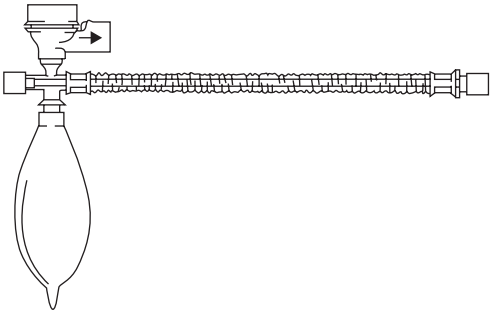

Fig. 25.2 Bain.

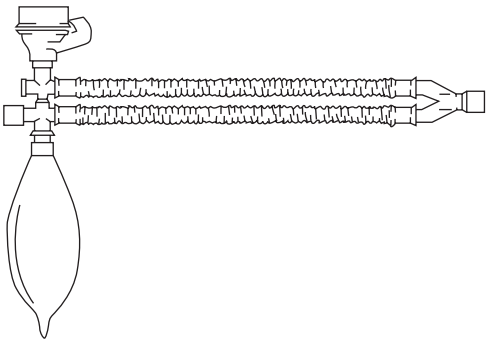

Fig. 25.3 Lack.

of oxygen through the tubing. If there are no leaks in the system, the oxygen bobbin will dip and the machine pressure relief valve will be activated. A flow rate of 2.5–3 times the minute volume should be used in this system to prevent rebreathing.

Lack (modified Mapleson 'A')

This is available as a coaxial or parallel system. Coaxial simply means one tube inside the other. These systems were developed to help control atmospheric pollution. The expiratory valve is placed at the anaesthetic machine end of the system, which facilitates easier scavenging of waste gases (Fig. 25.3). Fresh anaesthetic gases pass down the outer corrugated tube to the patient and, in a similar way to the Magill, some of the expired gases pass back up this tube. During the respiratory pause, the fresh gas supply pushes the last part of expired gas out of the valve via the inner expiratory tube. The position of the valve at the anaesthetic machine end of the system facilitates surgery on the head and makes this a less bulky system than the Magill. The Lack is not suitable for prolonged IPPV. A flow rate of 1–1.5 times the minute volume is recommended to prevent rebreathing.

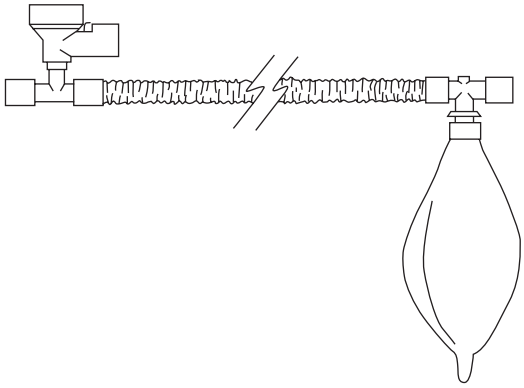

Fig. 25.4 Magill.

Mini Lack

The mini Lack is designed for patients less than 10 kg. It is more economical on gas than an Ayres T-piece as it can be used at 200 ml/kg. It is not suitable for IPPV.

Magill (Mapleson 'A')

The patient inhales fresh gas from the wide-bore corrugated tubing and reservoir bag, which are on the proximal end of the system (Fig. 25.4). Some of the exhaled gases pass back up this tubing, but, during the expiratory pause, the continuous fresh gas flow forces the last part of the expired gas (rich in carbon dioxide) out through a Heidebrink or APL valve into the scavenging system. A flow rate of at least the same as or slightly more than the patient's minute volume is required to prevent rebreathing. There is slight resistance to breathing due to the presence of the valve and this makes this system unsuitable for use in animals less than 5 kg. There is no upper weight limit for this system. The main disadvantage of this system is that it is not satisfactory for prolonged IPPV (expired gas is forced back into the lungs when the bag is squeezed instead of being removed via the expiratory valve, resulting in patient hypercapnia and hypoxia). The valve is situated on the distal end of the circuit making the circuit clumsy when used during head surgery.

REBREATHING SYSTEMS

- Circle.
- To and fro (rarely used in practice now).

These systems work in a completely different way to the non-rebreathing systems and, as their name suggests, exhaled gas is rebreathed after passing

through a canister of soda-lime granules (90% calcium hydroxide, 5% sodium hydroxide and 5% silicate), which remove the carbon dioxide. These systems use far less anaesthetic gas and volatile agent and produce less atmospheric pollution. These systems can be run completely closed at very low flow rates, or slightly higher gas flow rates can be used and excess gas is passed out into the atmosphere via an overflow valve. These systems have high resistance because of the packed soda lime and should only be used in animals over 10 kg. They are the most suitable and economic systems for use in large animal anaesthesia.

Frequent observation of the colour of the soda-lime granules is necessary to assess if they have become exhausted. It must be noted that colour change is no guarantee that the soda-lime is not exhausted. A more accurate assessment can be made by wrapping a small quantity in some gauze and blowing over it. The soda lime should heat up. If it does not it indicates that the soda-lime is exhausted. The VN must also be aware that different makes of soda-lime have different colour changes from fresh to exhausted, e.g. pink to white or white to pink or lilac.

Nitrous oxide should not be used with these systems unless an oxygen meter/pulse oximeter is used throughout the anaesthetic. This is due to the high concentration of nitrous oxide in the exhaled gas building up in the system and resulting in patient hypoxia.

There are two important factors to be aware of when using rebreathing systems.

- Following connection the concentration of the volatile agent is greatly reduced because it is mixed with the exhaled gas. Equilibrium occurs as soon as a certain blood concentration of anaesthetic gases has been reached, and gaseous agents are then exhaled from the body relatively unchanged.
- This first mixture of exhaled gas also contains nitrogen and this decreases the oxygen concentration of inhaled gas.

To overcome these problems, the circle and to and fro are best run as non-rebreathing systems until the patient is at the required level of anaesthesia. This means increasing the flow rate and opening the over-spill valve.

It should also be remembered that during anaesthesia the concentration of volatile agent within the system will progressively increase and the amount the patient inhales will not resemble what is dialled on the vaporiser. The oxygen consumption in the dog is 4–5 ml/kg/min. In theory this is all that is required to be set when using a to and fro or circle system; however, due to nitrogen build up in the system, a flow rate of 0.5–2 l/min is advisable.

As a general rule, it is advisable always to use these systems with the valve open, and not to use them as totally closed circuits. For the first 5–10 minutes, they should be run at a flow rate of about 3 l which is then reduced to 1 l for the remainder of the anaesthetic time.

The circle system

The soda-lime canister is placed in the vertical position far away from the patient's head (Fig. 25.5). This means that soda-lime inhalation, hyperthermia

Fig. 25.5 Circle system.

Table 25.2 Flow rates for Humphrey ADE.

Animal	Fresh gas flow recommendations (after induction) – minimum 300 ml/min
Cats – all weights	70–100 ml/kg/min, semi-closed, without absorber
Dogs <10 kg	70–100 ml/kg/min, semi-closed, without absorber
Dogs >10 kg	30 ml/kg/min induction; re-cycling with soda-lime

and an increase in dead space throughout anaesthesia are avoided. The inspiratory and expiratory tubes are fitted with unidirectional valves to ensure that the gases pass only in one direction. The main disadvantage of this system is that the combination of soda-lime canister and the valves make this a high-resistance system suitable only for animals over 10 kg.

HUMPHREY ADE

The Humphrey ADE breathing sytem combines the Mapleson A (Magill, Lack) and D (Bain) and E (Ayres T-piece). It can also incorporate a carbon dioxide absorber allowing gases to be recycled. It allows low fresh gas flow rates due to the special pop-off valve (Table 25.2). It is suitable for use for animals of weight range 6–90 kg. It allows spontaneous and controlled ventilation.

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SECTION 6

DIAGNOSTIC IMAGING

INTRODUCTION

The VN should be able to produce radiographs of diagnostic quality and, in order to do this, should have an understanding of the processes involved in producing the radiograph. This section covers the essentials of safe radiography and how to position the animal for common views. Preparation for contrast studies is covered in the final chapter, which also gives an overview of other techniques.

Chapter 26

Practical Radiography

DANGERS OF RADIOGRAPHY

Knowledge of the effects of ionising radiation is essential before undertaking any radiographic procedures. The potential dangers of radiation are often underestimated because:

- they are invisible;
- they are painless;
- their effects are latent (may take years to become apparent);
- the effects are cumulative – repeated low doses have an additive effect.

Effects of radiation injury

Radiation damages living tissues by changing biological molecules. Even low doses can cause cell damage because of the effect of radiation on DNA.

Somatic damage and carcinogenic effects

Damage that occurs to the body during the recipient's lifetime is called somatic damage. Direct changes in the body tissues, such as erythema, alopecia and digestive disturbances may occur soon after exposure, although latent affects may not be seen until many years later. Such disorders include cancer, cataracts, infertility and leukaemia.

Genetic damage

Damage can also occur to the reproductive cells, which can cause genetic mutations. These changes to the chromosomal material in the reproductive cells produce changes that are not apparent until future generations are born.

GENERAL SAFETY MEASURES

There are several safety measures that should be followed when undertaking radiography. Ionising radiation should only be used when there is a clear justification for carrying out the procedure. This will help reduce the amount of radiation produced in the practice. The ALARP principle states that a*ny* exposure of personnel to ionising radiation should be **As Low As Reasonably Practicable** (ALARP).

Distance

The intensity of radiation decreases with distance in accordance with the inverse square law. Never be any closer to the primary beam than 2 m.

Good radiographic technique

These steps will help to reduce risks.

- No part of any personnel should ever be within the primary beam.
- Collimate to the area of interest.
- Use fast film–screen combinations when possible to reduce exposure.
- Avoid repeat exposures by using good positioning methods, exposure charts and standardised processing techniques.
- Avoid using a grid if it is not needed as grids will require a higher exposure.
- Use positioning aids/anaesthesia to avoid holding animals.

Premises

- Identify the controlled area (2 m around the primary beam).
- Walls 15 cm concrete or double-brick.
- Lead ply or barium plaster.
- Lead screens. Lead-backed table.
- Warning notices/lights (lit when exposure taken).
- International radiation symbol displayed on entry to the room.

Equipment

- Machines need to be checked and serviced regularly.
- Check the accuracy of the light beam diaphragm.
- Check the integrity of lead aprons, gloves and screens.

Staff

- Limited access to controlled area.
- Only essential and trained personnel in the controlled area.
- No one under 16 years old in the controlled area.
- No pregnant women in the controlled area.
- Limited access to controlled area by 16–18-year-old personnel.
- Monitor personnel using dosimeters.

Protective clothing

Lead clothing protects against scatter but not against primary radiation.

- Lead aprons (>0.25 mm lead).
- Lead gloves and hand shields (>0.35 mm lead).

- Lead thyroid shields.
- Lead glasses (ocular tissue is one of the most delicate areas subjected to radiation exposure, yet the eyes are most frequently neglected).

Ensure the equipment is stored correctly and not folded.

Chapter 27

Selection of Exposure Factors

EXPOSURE FACTORS

The exposure factors that are selected have a huge impact on how the finished film will turn out. Generally, the kilovoltage (kV) should be increased as tissue thickness and density increase. The milliamperage (mA) is usually left the same. Provided that processing techniques are adequate:

- dark images are overpenetrated so the kV should be reduced;
- white, pale images are underpenetrated so the kV should be increased.

Kilovoltage

Increasing the kV will increase the penetrating ability of the X-ray photons and therefore the effect on the film is to produce blacker images of lower contrast. The contrast of the film is the difference between the white areas and the black areas – high contrast means there are definite areas of black and white and all the greys in between. Low contrast means that there is a general all over greyness.

Milliamperage and time

The amount of X-rays produced multiplied by the time gives the milliampere–seconds (mAs). Increasing mAs will produce more X-ray photons. Changing the mAs does not affect the penetrating power of the X-ray photons but it will produce a blacker image on the areas of film that have been penetrated. The relationship between the milliamperes and the seconds allows a range of exposures to be used depending on the machine.

Distance (film–focal distance)

The distance between the focal spot (in the tube head) and the film affects the quantity of X-rays reaching the film. The greater the distance is, the more the beam spreads out therefore fewer X-rays reach the film. Usually the film–focal distance (FFD) is kept constant, between 75 cm and 100 cm. A less powerful machine would require a shorter distance. If the FFD is increased, the intensity of the beam decreases according to the inverse square law. Therefore, if

Table 27.1 Example of an exposure chart.

	Area	mA	kV	Time (sec)	FFD	Film	Grid
Dog – giant	Lateral stifle	100	90	0.6	75 cm	Fugi G8 18 × 24	Yes
Dog – large	Lateral stifle	100	85	0.2	75 cm	Fugi G8 18 × 24	No
Dog – medium	Lateral stifle	100	80	0.08	75 cm	Fugi G8 18 × 24	No
Dog – small	Lateral stifle	100	65	0.02	75 cm	Fugi G8 18 × 24	No
Cat	Lateral stifle	100	60	0.02	75 cm	Fugi G8 18 × 24	No

the X-ray tube head is too far away from the object being radiographed, the image will be overly faint or non-existent.

Exposure charts

An exposure chart provides a quick, visual display of all of the exposures that are needed for each body area (Table 27.1). The aim of an exposure chart is to avoid repeating exposures, to save time and wasted films and to reduce the radiation hazard to staff. Settings can be chosen by assessing the size of the patient, i.e. cat, small dog, medium dog and large dog. Be aware that charts apply only to a particular X-ray machine, using constant FFDs, cassettes, films, grid and processing technique.

Other factors that should be taken into account when setting the exposure are:

- movement: as short a time as possible should be selected (movement can be from respiration);
- dressings and casts: the kV must be increased by 10 for casts.

Variations from the exposure chart are sometimes necessary because of the limitations of the machinery, limitations of the environment or limitations created by the patient.

EXPOSURE CALCULATIONS

Film–focal distance and the inverse square law

Occasionally, it may be necessary to alter the FFD. For example, if there is a low-output X-ray machine, and it is necessary to penetrate thicker tissue than the machine is capable of at its usual FFD (e.g. large animal radiography), it is possible to decrease the FFD in order to achieve penetration and a radiograph of reasonable quality. If the FFD is altered, the new exposure can be worked out using this calculation:

$$\text{New mAs} = \text{New FFD}^2 \div \text{Old FFD}^2 \times \text{Old mAs}$$

Example

Having been using an exposure of 10 mAs and FFD of 50 cm, what exposure is required if the FFD is changed to 100 cm?

$$\text{Answer: } 10 \times 10000 \div 2500 = 10 \times 4 = 40 \text{ mAs.}$$

The 10 kV rule

Increasing the kV by 10, does the same (more or less) to the image as doubling the mAs. To put this another way, doubling the mAs does the same to the image as increasing the kV by 10. Likewise, decreasing the kV by 10 does the same to the image as halving the mAs would.

Why is this useful?

It is possible to use this rule in all kinds of situations when the machine or patient is causing problems. For example, the patient is panting and the rapid breathing movement is creating a very badly blurred image on the finished radiograph. The exposure settings produced an adequate image, but it is necessary to reduce the exposure *time* to prevent the movement blur. Another situation where this calculation would be useful is when a darker image is required but the kV is up as high as it will go. Using the 10 kVp rule to keep the kV as it is but doubling the mAs gives the darkening required.

Milliamperage and time

This has already been covered in this chapter, but as a reminder:

$$\text{mAs} = \text{mA} \times \text{s}$$

If mA is doubled, the time may be halved (thereby decreasing exposure time and reducing the possibility of movement blur).

Grid factor

Grids are used to improve the quality of the finished radiography when radiographing through thicker body parts. They also help to absorb scattered radiation. When using a grid it is necessary to increase the exposure by something called the *grid factor*:

$$\text{mAs} \times \text{grid factor (GF)} = \text{exposure required using grid}$$

Example

The exposure factors required for a lateral radiograph of a dog's abdomen might be 3.2 mAs at 80 kV. If a grid with a grid factor of 3 is used, the new exposure will be 3.2 × 3 = 9.6 mAs at 80 kV. This exposure time may be too long, so the 10 kV rule can be used. By increasing the kV by 10 it is possible to halve the mAs, giving a setting of 90 kV at 4.8 mAs.

Chapter 28

Radiographic Positioning

The VN should be able to position an animal correctly in order to produce a diagnostic radiograph of any part of the animal. To describe a radiographic projection, the point of entry of the beam is first described followed by the point of exit. For example, dorsoventral thorax means that the animal is in sternal recumbency and the X-ray beam enters dorsally through the spine and exits ventrally though the sternum.

The following are terms used to describe radiographic projections:

- cranial;
- caudal;
- rostral;
- dorsal;
- ventral;
- proximal;
- distal;
- medial;
- lateral;
- palmar;
- plantar.

The terms *anterior* (towards the head) and *posterior* (towards the tail) are sometimes used instead of cranial and caudal (Fig. 28.1).

RESTRAINT

Effective restraint is necessary not only to prevent the animal from moving, but also to help position the area being radiographed so that the best possible view can be achieved. Restraint can be achieved in two ways:

- chemically: sedation or general anaesthesia;
- positioning aids: ties, sandbags, foam wedges, radiolucent troughs, Velcro™.

An animal must *never* be held during radiography. Manual restraint is exceedingly dangerous and should only ever be used in exceptional circumstances, in which case the requesting veterinary surgeon should restrain the animal. The VN must *never* do this. Under the Ionising Radiation Regulations 1999 it is totally unacceptable to hold animals. Never use ties to secure body

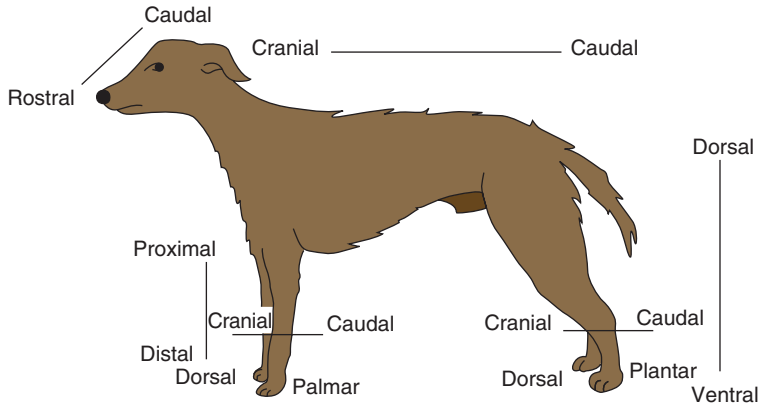


Fig. 28.1 Anatomical directions.

areas on conscious or sedated animals. Serious damage and distress can occur if they are roused and try to move.

ANATOMICAL KNOWLEDGE

Thorough knowledge of anatomy and bony landmarks is essential to ensure good positioning of the animal for radiography. The VN must be fully competent in this, not only for written and practical examinations but also for the following reasons:

- ensures that all of the relevant area is on the radiograph;
- enables ease of interpretation of the final radiograph;
- standardises methods so that comparisons with books and other radiographs can be made;
- helps to prevent repeat radiographs being taken (safety, cost and time considerations);
- prevents other body structures obscuring the relevant area;
- ensures structures are not distorted by incorrect centring.

POSITIONING OF THE ANIMAL

Being able to produce consistently good radiographs is highly rewarding and an invaluable aid to the veterinary surgeon in the diagnosis of many conditions. To achieve this, it is necessary to standardise patient positioning, centring points and collimation areas. What follows illustrates the main positioning, centring points and collimation areas for most standard radiographic procedures. These are guidelines designed to help the VN in practice and also in examinations. It is important to remember that each case is different and the personal preferences of each veterinary surgeon should be considered.

Thorax

The exposure should be taken during peak inspiration so that the lungs are fully expanded and aerated. This helps to increase the contrast on the finished radiograph. Exposure *time* must be kept to a minimum to help reduce movement blur from respiration movement. In anaesthetised animals the lungs can be artificially inflated temporarily whilst the radiograph is taken. See Figs 28.2–28.4.

Abdomen

It is usually recommended that the animal be starved for at least 12 hours prior to abdominal radiography. Cleaning and brushing and drying the coat first will help to produce clearer pictures by removing confusing radiodensities. It is advisable to administer an enema before any radiographic studies of the urogenital tract and colon. The exposure should be taken during

Fig. 28.2 Lateral thorax.

Lateral thorax This position is useful for imaging the heart and lungs and checking for lung metastases	
Position	Place the animal in right or left lateral recumbency Extend the forelimbs cranially, secure with sandbags (or ties if anaesthetised) Extend the head and neck and secure with sandbag if necessary Place a foam wedge under sternum to help ensure sternum and spine are in the same horizontal plane
Centre on	Caudal aspect of the scapula, midway between dorsal edge of scapula and sternum
Collimation	(a) manubrium and last rib (b) lateral skin surfaces

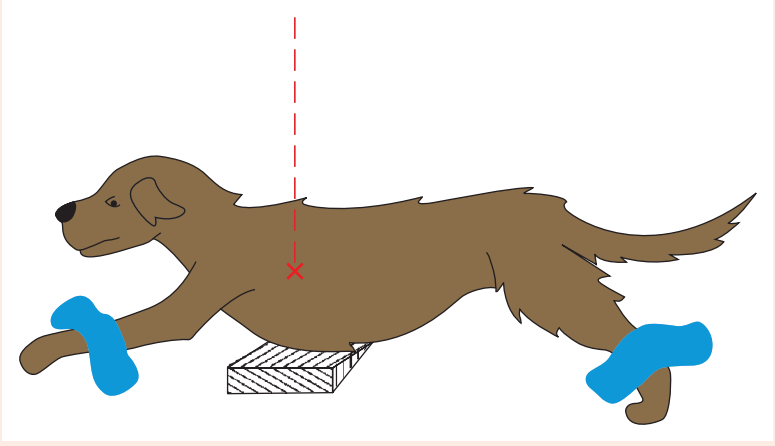


Fig. 28.3 Dorsoventral thorax.

Dorsoventral thorax This is useful for imaging the heart as it is closest to the cassette in this position	
Position	Sternal recumbency Forelimbs extended cranially, secured with sandbags or ties Sandbags either side of animal to help prevent lateral rotation of the body
Centre on	Midline at the level of the caudal scapula
Collimation	(a) manubrium and last rib (b) lateral skin surfaces

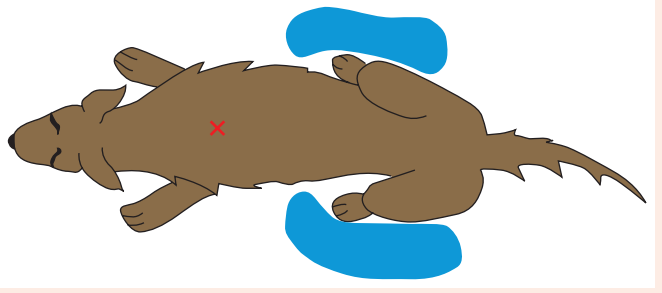
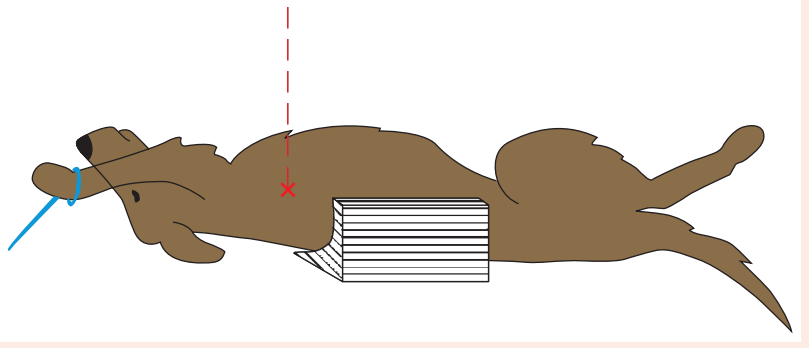


Fig. 28.4 Ventrodorsal thorax.

Ventrodorsal thorax This rarely carried out because the heart adopts a more natural position during dorsoventral (DV) views. Note: Avoid placing a dyspnoeic animal in dorsal recumbency for a ventrodorsal view of the thorax as this can exacerbate the problem. This particularly applies to an animal with a ruptured diaphragm	
Position	Dorsal recumbency Forelimbs extended cranially, secured with sandbags or ties Sandbags or trough to prevent lateral rotation of the body
Centre on	Midline at the level of the caudal scapula
Collimation	(a) manubrium and last rib (b) lateral skin surfaces



expiration. Centring and collimation of the primary beam is described below for general abdominal views. If specific organs and structures are required, centring of the primary beam will need to be altered to take this into consideration. For example, the beam will need to be centred further cranially for examinations of the liver. Bladder studies will require that the beam be centred further caudally. Good anatomical knowledge is required to be able to perform the required task appropriately. Dorsoventral views of the abdomen are rarely indicated. Exceptions would be for dyspnoeic patients and those having contrast studies of the stomach. See Figs 28.5 and 28.6.

Skull

Anaesthesia is required for all views of the skull to enable good positioning. If present the endotracheal tube should be temporarily removed during exposure, to avoid confusing artefacts on the finished radiograph. The views described below demonstrate the positioning, centring and collimation for general views of the skull. These may vary depending on the area of interest and good anatomical knowledge of the skull is required to obtain relevant radiographs. See Figs 28.7–28.12.

Fig. 28.5 Lateral abdomen.

Lateral abdomen	
Position	Animal in lateral recumbency (usually right lateral recumbency – be consistent) Hindlimbs extended caudally and secured with sandbags or ties A foam wedge may be placed between the stifles and under the sternum to help reduce horizontal rotation of the body
Centre on	Last rib, midway between dorsal and ventral skin surface (for general abdominal view).
Collimation	(a) just cranial to the xiphisternum and to the greater trochanter (b) the spine dorsally and the ventral skin surface

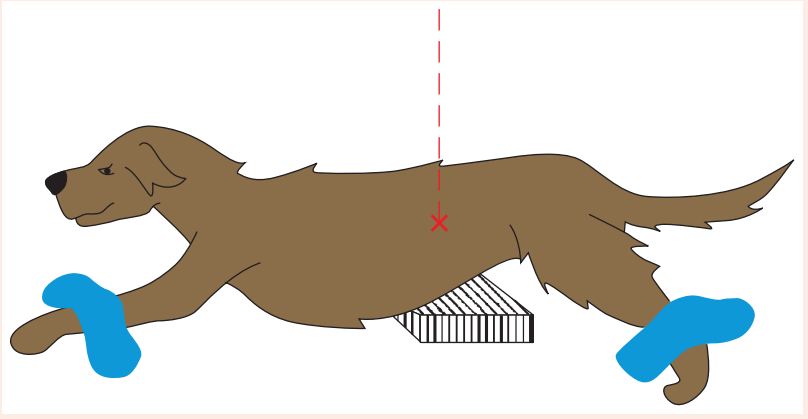


Fig. 28.6 Ventrodorsal abdomen.

Ventrodorsal abdomen	
Position	Animal in dorsal recumbency Sandbags or troughs to prevent lateral rotation of the body Hindlimbs extended caudally
Centre on	Midline, level with the last rib
Collimation	(a) just cranial to the xiphisternum and to the greater trochanter (b) the lateral skin edges

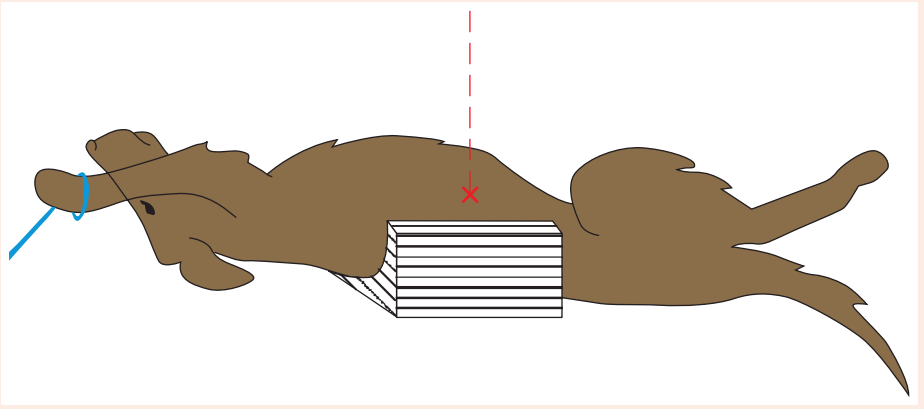


Fig. 28.7 Lateral skull.

Lateral skull	
Position	Animal in lateral recumbency Place small foam wedge under the muzzle to ensure that the sagittal plane of the skull is parallel to the cassette Extend neck
Centre on	Midway between the eye and the ear
Collimation	(a) external nares to caudal skull (b) top of skull to ventral aspect of mandible

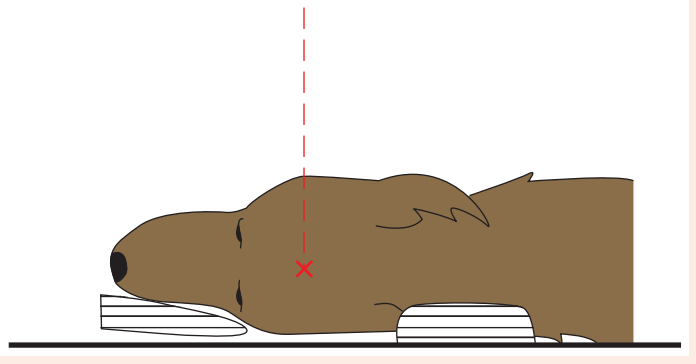


Fig. 28.8 Dorsoventral skull.

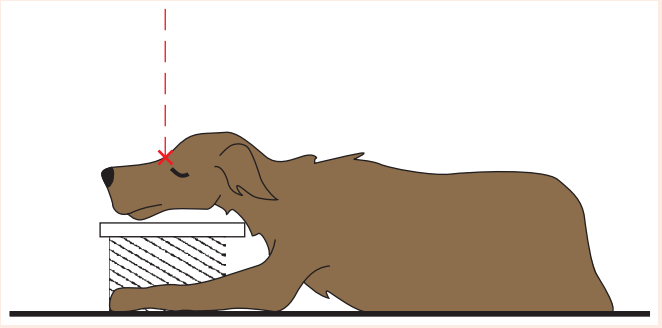
Dorsoventral skull	
Position	Animal in sternal recumbency, neck extended Place head and cassette on a foam block or other, to make sure entire skull is parallel to the cassette Place sandbag over neck to help prevent lateral rotation
Centre on	Midline, level with the medial canthus of the eye
Collimation	(a) external nares to caudal edge of skull (b) lateral skin surfaces
	

Fig. 28.9 Ventrodorsal skull.

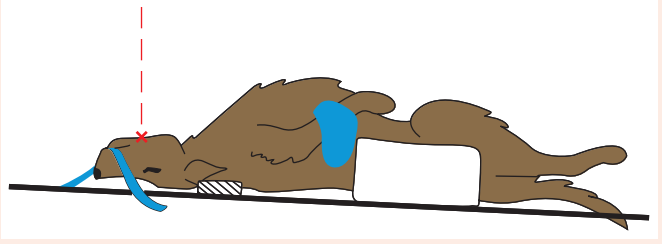
Ventrodorsal skull	
Position	Animal in trough in dorsal recumbency, neck extended Place foam pad under the neck to help ensure palate is parallel to the cassette Place adhesive tape around the upper canines and fixed to the table top or cassette
Centre on	Midline, level with the medial canthus of the eye
Collimation	(a) external nares to caudal skull (b) lateral skin surfaces
	

Fig. 28.10 Rostrocaudal skull (frontal sinuses).

Rostrocaudal skull This view is performed to evaluate the frontal sinuses, temporal area and zygomatic arch.	
Position	Animal in trough in dorsal recumbency Limbs secured caudally using sandbags to help prevent lateral rotation Ties applied around the muzzle so that the skull is perpendicular to the cassette
Centre on	Frontal sinuses
Collimation	(a) lateral sides of skull (b) top of skull and nares

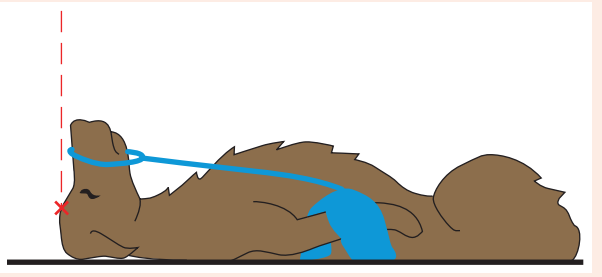


Fig. 28.11 Open-mouth rostrocaudal skull.

Open-mouth rostrocaudal An 'open-mouth' rostrocaudal view is obtained mainly to evaluate the tympanic bullae	
Position	As before, placing a tie around maxilla and secured to the table to ensure skull and hard palate are perpendicular to the cassette Pull the mouth open and place a tie around the mandible and secure to the table Pull the tongue out of the mouth as much as possible
Centre on	Direct primary beam at a 5–10° angle to the vertical. Centre on back of larynx
Collimation	(a) midline, between the eyes (bulla – midline, base of the tongue) (b) the skin surfaces around the skull

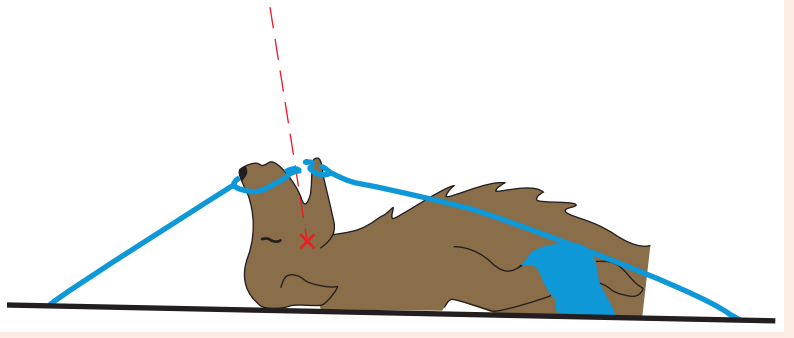
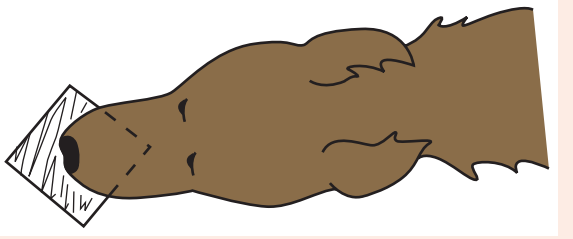


Fig. 28.12 Intraoral.

Intraoral This view is used chiefly to evaluate the teeth and nasal cavity	
Position	Animal in sternal recumbency Troughs or sandbags to prevent lateral rotation Muzzle placed on a foam block Flexible cassette with non-screen film placed in the mouth over the tongue
Centre on	Midline halfway along muzzle – depends on area of interest
Collimation	Include nasal chambers from the external nares to caudal oral cavity



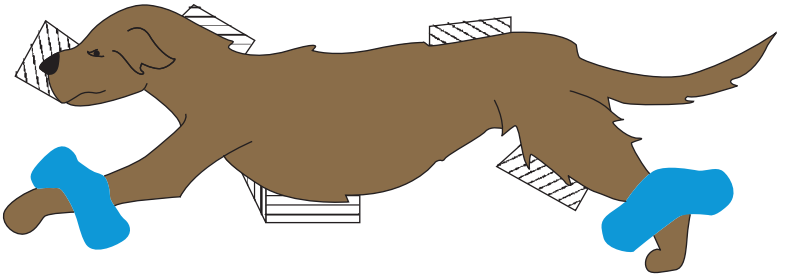


Fig. 28.13 Lateral spine.

Spine

In order to get accurate radiographs of the spine it should be radiographed in three sections: cervical, thoracic and lumbar. For lateral views it is essential to ensure the spine lies in a horizontal plane to the table (Fig. 28.13). The disc spaces should be perpendicular to the table (Fig. 28.14). Foam pads are placed under mandible, cervical and lumbar spine. A foam wedge under the sternum and between the legs helps to prevent rotation of the body. The positioning is important to ensure there is no distortion in the final radiograph. See Fig. 28.15.

Lateral cervical spine	
Position	Patient in lateral recumbency Forelimbs drawn caudally and secured
Centre on	C4–C5 vertebrae
Collimation	(a) C1 to cranial edge of scapula (b) dorsoventrally to include muscle mass but not fat and skin
Lateral thoracic spine	
Position	Patient in lateral recumbency Forelimbs extended cranially and hindlimbs extended caudally
Centre on	Seventh thoracic vertebra
Collimation	(a) cranial scapula to the last rib (b) dorsal skin surface to mid thorax
Lateral thoracolumbar spine	
Position	Patient in lateral recumbency Forelimbs extended cranially and hindlimbs extended caudally
Centre on	The thoracolumbar junction
Collimation	(a) three vertebrae cranial and caudal to the thoracolumbar junction (b) dorsal skin surface to mid thorax/abdomen
Lateral lumbar spine	
Position	Patient in lateral recumbency Forelimbs extended cranially and hindlimbs extended caudally
Centre on	Fourth lumbar vertebrae
Collimation	(a) from 13th thoracic vertebra to first sacral vertebra (b) dorsal skin surface to mid abdomen

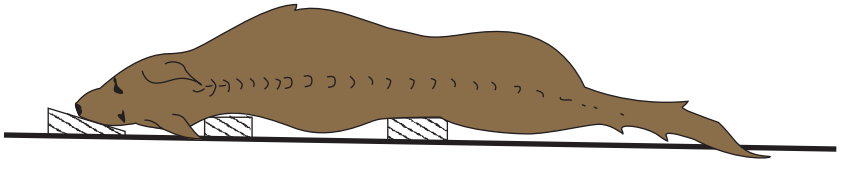


Fig. 28.14 Lateral spine (foam wedges).

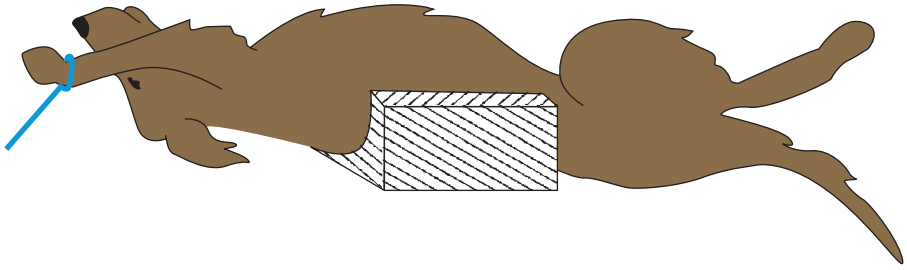


Fig. 28.15 Ventrodorsal spine.

The ventrodorsal views of the spine should also be taken in sections. It is important to ensure the spine is straight – this may be achieved using slight traction with ties on fore- and hindlimbs. Foam pads are necessary to prevent rotation or the animal is placed in a trough.

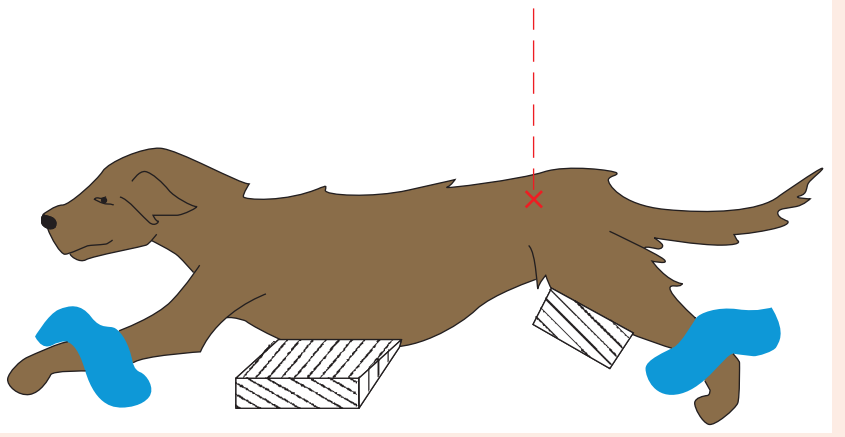
Ventrodorsal cervical spine	
Position	Animal placed into a trough in dorsal recumbency Forelimbs drawn caudally and secured
Centre on	Midway between the occipital crest and the caudal aspect of the scapula over C4–C5 vertebrae
Collimation	(a) occipital crest and caudal aspect of the scapula (b) lateral skin surfaces
Ventrodorsal thoracic spine	
Position	Animal placed into a trough in dorsal recumbency Forelimbs drawn cranially and secured
Centre on	Level with the caudal aspect of the scapula over the vertebrae (T6)
Collimation	(a) cranial scapula to the last rib (b) lateral skin surfaces
Ventrodorsal thoracolumbar spine	
Position	Animal placed into a trough in dorsal recumbency Forelimbs drawn cranially and secured
Centre on	The thoracolumbar junction
Collimation	(a) three vertebrae cranial and caudal to the thoracolumbar junction (b) lateral skin surfaces

Continued

Ventrodorsal lumbar spine	
Position	Animal placed into a trough in dorsal recumbency Forelimbs drawn cranially and secured
Centre on	Fourth lumbar vertebra
Collimation	(a) from 13th thoracic vertebra to first sacral vertebra (b) lateral skin surfaces

Fig. 28.16 Lateral pelvis.

Lateral pelvis	
Position	Patient in lateral recumbency with affected side closest to cassette. The limb closest to the cassette should be pulled slightly cranially whilst the upper limb is pulled caudally Foam wedges between stifles and under sternum to prevent rotation of the body
Centre on	The greater trochanter of the femur
Collimation	(a) the iliac crest and ischial tuberosity (b) the dorsal skin surface and mid femur

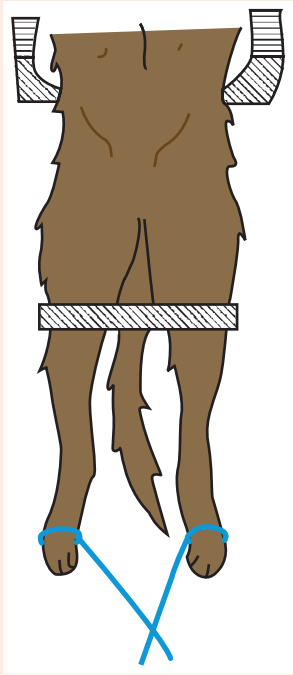


Pelvis

A foam wedge is placed between the stifles on the lateral view of the pelvis to prevent rotation (Fig. 28.16). The aim is to ensure the hindlimbs are horizontal to the table. The ventrodorsal view may be a relaxed 'frog-leg' view or with the hindlimbs extended as for a hip dysplasia survey film (Fig. 28.17).

Fig. 28.17 Ventrodorsal pelvis.

Ventrodorsal pelvis – extended	
Position	Patient in trough, in dorsal recumbency Hindlimbs extended caudally and fixed with ties to the table Legs rotated medially to ensure femurs are parallel to each other. Palpate the patellae to ensure they are pointing upwards Secure in the correct position around femurs with adhesive tape or 'Velcro'
Centre on	Midline, level with the greater femoral trochanters
Collimation	(a) iliac crest and patellae (b) lateral skin surfaces The patellae must be included in the finished radiograph when being submitted for BVA/KC hip score



Forelimb

It may be necessary to take comparative films of both limbs especially in the case of fractures to check for limb shortening from contraction of the muscles.

Shoulder

The lateral shoulder position is often referred to as the 'superman' pose (Fig. 28.18). This should help you remember! See also Fig. 28.19.

Fig. 28.18 Lateral shoulder.

Lateral shoulder	
Position	Animal in lateral recumbency on the same side as the shoulder being radiographed. Extend the head slightly Extend the required limb cranially and secure with tie. Draw the other limb caudally and secure
Centre on	The shoulder joint
Collimation	(a) midway along the scapula and proximal third of the humerus (b) approximately 4–5 cm cranial and caudal to the greater tuberosity If a radiograph of the scapula is required, centre the beam midway between the greater tuberosity and the dorsocaudal aspect of the scapula

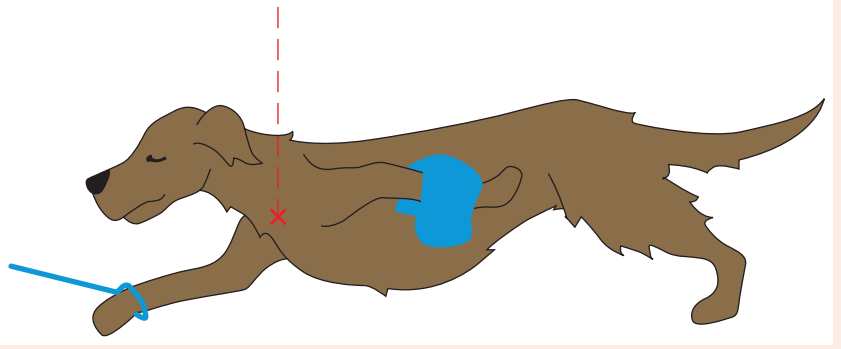
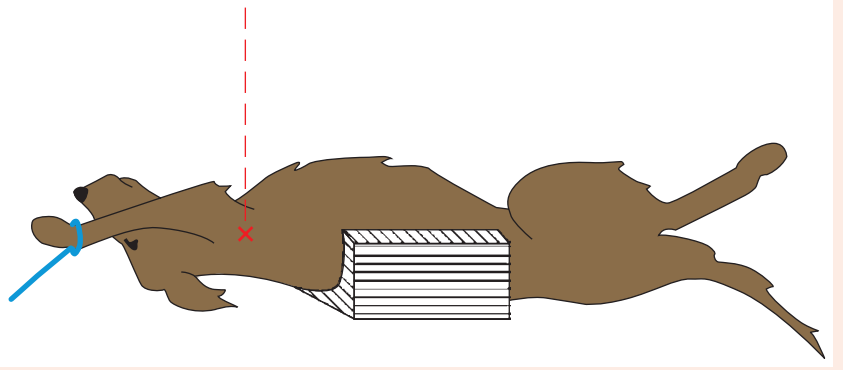


Fig. 28.19 Caudocranial shoulder.

Caudocranial shoulder	
Position	Animal in trough, in dorsal recumbency Extend limb of interest cranially and secure with tie to the table
Centre on	Shoulder joint
Collimation	(a) midway along the scapula and proximal third of the humerus (b) lateral skin surface and midline thorax



Humerus

The animal is in the same position as for the shoulder. The body may require slightly more rotation to expose the lower humerus for the lateral view. A craniocaudal view may be taken with the animal in dorsal recumbency and the affected limb pulled caudally and tied. The distance between the cassette and limb will produce some degree of magnification.

Lateral humerus	
Position	Position animal the same way as for the lateral shoulder
Centre on	Midway between shoulder joint proximally and elbow distally, on the bone
Collimation	Include both the shoulder and elbow joint and lateral skin surfaces
Caudocranial humerus	
Position	Position the animal the same way as for the caudocranial shoulder
Centre on	Midway between shoulder joint proximally and elbow distally, on the bone
Collimation	(a) include both the shoulder and elbow joint (b) lateral skin surface and midline thorax

Elbow

Fully flexed lateral views are required for elbow dysplasia screening. For a craniocaudal view the beam may need to be angled slightly into the joint. Take care with arthritic joints. See Figs 28.20 and 28.21.

Radius/ulna

See Fig. 28.22.

Carpus and foot

Lateral and dorsopalmar carpus and foot	
Position	Position same as for radius/ulna If a radiograph of the individual phalanges or pad is required, the toes can be spread apart with tape and a lateral view obtained
Centre on	The carpus or middle of foot depending on area of interest
Collimation	(a) just proximal to carpus to end of digits (b) include the lateral skin surfaces

Fig. 28.20 Lateral elbow.

Lateral elbow	
Position	Position the animal in the same way as for the shoulder and humerus but ensure the elbow is either in a natural flexed position or fully flexed Some veterinary surgeons prefer the joint to be fully flexed; others favour a more neutral position
Centre on	Middle of medial humeral condyle
Collimation	One third of the way along the humerus proximally and one third of the way along the radius and ulna distally

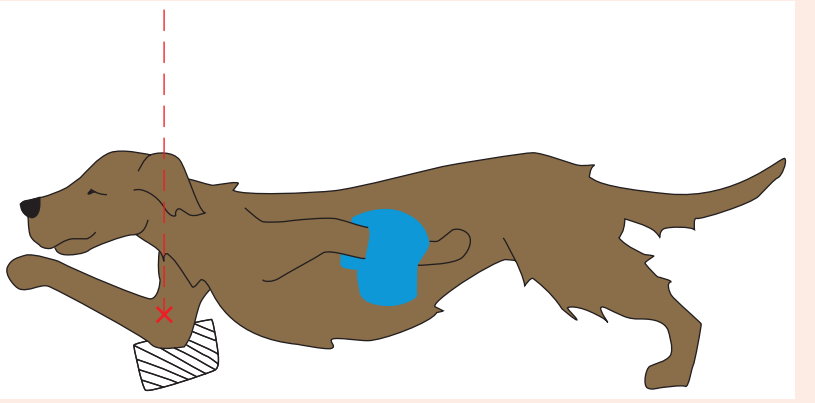


Fig. 28.21 Craniocaudal elbow.

Craniocaudal elbow	
Position	Animal in sternal recumbency Draw limb of interest cranially and secure to table with tie. Place foam pad in between the elbow and the body to prevent rotation
Centre on	Midline, level with the humeral condyles
Collimation	(a) one third of the way along the humerus proximally and one third of the way along the radius and ulna distally (b) Lateral skin surfaces

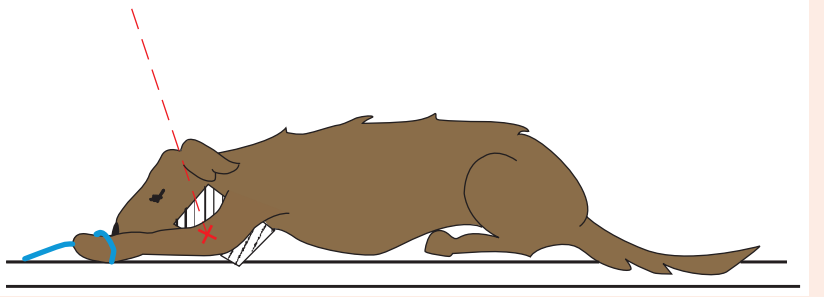
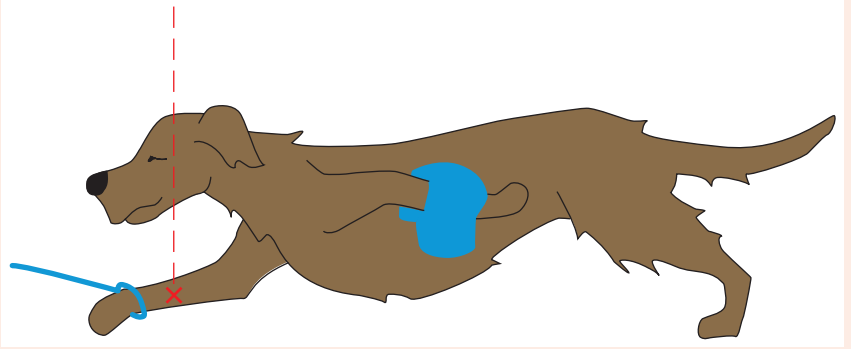


Fig 28.22 Lateral radius/ulna.

Lateral and craniocaudal radius/ulna	
Position	Both the lateral and craniocaudal views can be positioned in the same way as the elbow
Centre on	Midway between elbow and carpus
Collimation	Include the elbow and the carpus joints



Hindlimbs

Femur

See Fig. 28.23 and Fig. 28.24.

Stifle

See Fig. 28.25. The caudocranial view may also be taken with the animal in dorsal recumbency. A foam pad is placed under the opposite hindlimb to ensure the stifle is not rotated. The lateral view is the same for lateral femur.

Tibia and fibula

Lateral and craniocaudal tibia/fibula	
Position	Position animal in the same way as for the femur and stifle
Centre on	Midway between tarsus and stifle
Collimation	Include the stifle and hock joints and lateral skin surfaces

Hock and foot

Lateral and dorsoplantar hock and foot	
Position	Position in the same way as femur, stifle and tibia/fibula
Centre on	The tarsus or foot depending on the area of interest
Collimation	(a) just proximal to hock to end of toes (b) lateral skin surfaces

Fig. 28.23 Lateral femur.

Lateral femur	
Position	Animal in dorsal recumbency but turned slightly so that the limb of interest falls towards the table Place sandbags around the body to maintain the correct position Extend the other limb and secure away from limb of interest Place small foam wedge under hock to ensure femur is parallel to the cassette
Centre on	Midway along femur between stifle and greater trochanter
Collimation	Include the hip and stifle joints

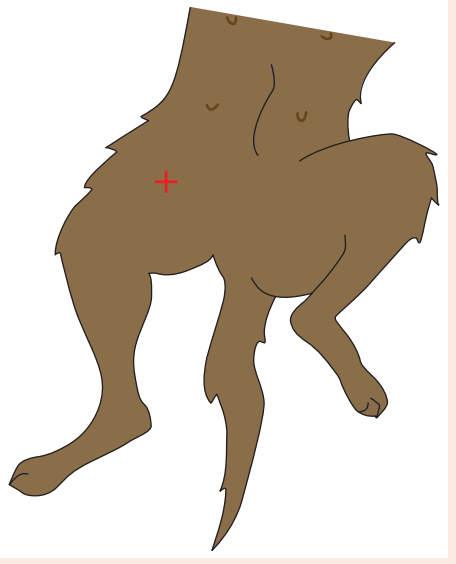


Fig. 28.24 Craniocaudal femur.

Craniocaudal femur	
Position	Animal in trough, in dorsal recumbency Extend limb of interest and secure using tie and sandbag Secure other limb out of the way
Centre on	Midway along femur between stifle and greater trochanter
Collimation	Include the hip and stifle joints

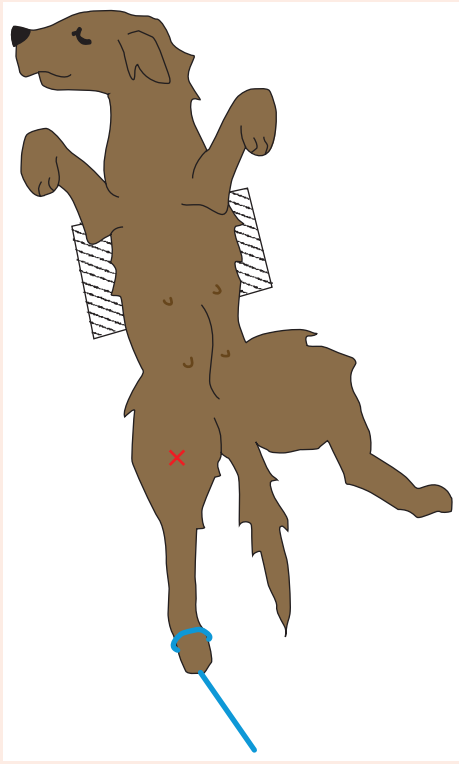


Fig. 28.25 Craniocaudal stifle.

Craniocaudal stifle	
Position	Position animal in the same way as for the femur
Centre on	Just distal to the femoral condyles
Collimation	Include the proximal one third of the femur and distal one third of tibia/fibula

Chapter 29

The Processing of Radiographs

Although many practices now have automatic processors it is still important that nurses are able to manually process films and understand the procedure involved.

Radiographs may be processed by two different methods:

- manually with tanks (wet processing);
- automatically using processor machines (dry processing).

PROCESSING PROCEDURE

The processing procedure consists of five stages.

(1) Development.
(2) Rinsing (omitted in automatic processing).
(3) Fixing.
(4) Washing.
(5) Drying.

Processing must be carried out in the dark, under safelight conditions because the X-ray film is very sensitive to light before it is exposed. Exposure to white light before processing causes the film to turn entirely black.

Development

During development, the *exposed* silver bromide crystals in the emulsion are 'reduced' or converted in to grains of *black* metallic silver.

Rinsing

Rinsing the film removes surplus developing chemicals to prevent contamination of the fixer solution. The film should be placed in the rinse tank for about 10 seconds. In automatic processors, this stage is replaced by the film going through rollers to squeeze excess developer solution off the film.

Fixing

Fixing of the film is required for several reasons:

- the film is still sensitive to white light and must be 'fixed';
- it prevents the film from continuing to be developed;
- it removes the unexposed silver bromide crystals;
- it hardens the emulsion;
- it makes the image permanent.

The fixer becomes exhausted due to the build up of silver bromide ions. Exhaustion happens quicker to fixer than it does to developer. The speed of fixing increases with high temperatures and with agitation. Fixer temperature is not critical as long as it remains below 21°C; above this, temperature staining may occur.

Washing

Washing is essential to remove the chemicals from the film and prevent discoloration of the film in storage. The film should be rinsed under running water for at least 15–30 minutes. Hard water may produce 'scum' on film and water softeners are often incorporated into automatic processors. A 'wetting agent' may be used before drying to ensure the film is evenly wet, reducing the possibility of drying marks. In automatic processors the film passes through the rinse tank to remove excess fixer solution and is squeezed dry by the rollers.

THE MANUAL PROCESSING PROCEDURE

(1) Check the developer and fixer levels.
(2) Check the developer temperature (20°C, 68°F) and stir.
(3) Ensure that hands are clean and dry.
(4) Switch on the safelight and load the film hanger with the exposed film.
(5) Place the film in the developer, agitate slowly and gently, and replace the lid to prevent oxidation of the developer solution.
(6) Develop for 3–5 minutes.
(7) Remove the film from the developer and drain (over the developer tank) to remove excess liquid from the film and hanger.
(8) Rinse film immediately before putting into fixer.
(9) Place the film in the fixer, after 30 seconds the light may be switched on to check the film. Then fix for a further 10 minutes.
(10) Remove the film from the fixer and rinse for 15–30 minutes in running water.
(11) Remove the film from the hanger and allow to dry in a dust-free environment.

AUTOMATIC PROCESSORS

Automatic processors only have three tanks: the developer, the fixer and washer dryer. The films pass through a system of rollers in the machine. The rinsing phase between the developer and fixer is omitted due to the

'squeegee' effect of the rollers. In the wash tank, the water flows at a rate of around 6l/min so that any contaminants are washed away. The developer and fixer are stored in reservoir tanks and measured quantities are pumped into the appropriate tanks. Replenishment rate of the chemicals is adjusted to suit film throughput, i.e. the more films that are developed the more replenishing takes place.

(1) Ensure the machine is switched on for about 10 minutes prior to use, to warm up (this may be longer in cold weather).
(2) Use an old, clean film to pass through the processor after the warm-up period to ensure the machine is working and to remove dried-on chemicals from the rollers.
(3) In the darkroom with the safelight on remove the exposed film from the cassette and feed gently into the processor.
(4) Load the empty cassette with new film and close it firmly.
(5) Ensure all of the film has disappeared into the processor before switching the light on.

At the end of the day, the machine should be turned off and the rollers wiped clean. Automatic processors should be serviced regularly. The rollers and tanks need cleaning regularly. In most automatic processors, the rollers are easily removed for cleaning.

FILM STORAGE

Unexposed film is sensitive to light and must be stored in a light proof filmhopper or the original film box. Film boxes and loaded cassettes must be kept away from the X-ray area and X-ray film should also be kept away from:

- chemical fumes;
- pressure (always keep boxes of film upright);
- excessive humidity;
- high temperatures.

FILM IDENTIFICATION

All radiographs need to be labelled permanently with:

- patient identification;
- date;
- left/right marker;
- any other relevant details, e.g. time after contrast.

Labelling can be performed at any of these three stages:

(1) *During exposure.* Using lead letters or X-rite™ tape on the outside of the cassette. A left/right marker should also be used at this time.

(2) *After exposure.* In the darkroom using a white-light marker. A small piece of lead in the corner of the cassette protects the film from being exposed to X-ray photons. This corner can then be exposed to white light from behind a piece of paper with the relevant details written on it. This is not an acceptable method for British Veterinary Association/Kennel Club (BVA/KC) hip dysplasia radiographs.
(3) *After processing.* Using a chinograph pencil or adhesive label. This is not a good method and is not acceptable for legal cases.

Labelling films for BVA/KC hip and elbow scoring

Radiographs taken for submission to the BVA/KC must be labelled with:

- the dog's kennel club number (recent legislation recommends chip ID numbers or tattoos should also be required for indentification http://www.bva.co.uk/canine_health_schemes/Hip_Scheme.aspx);
- date;
- left/right marker.

Absolutely no other information is allowed. Acceptable methods of identification for this scheme are only those used at the time of exposure, i.e. lead letters or X-rite tape.

Chapter 30

Appraisal of the Film

To enable accurate interpretation of the finished film, it is essential that the correct area of the animal has been radiographed *and* that the finished radiographs are of a high diagnostic quality, with good contrast between the body structures and tissues. They must be of clear, sharp quality and have no misleading artefacts. This requires:

- correct exposure settings;
- good and standardised processing methods.

ASSESSING THE QUALITY OF THE RADIOGRAPH

When the film is developed, the VN should examine it for any of the faults below. The radiograph should always be viewed on a viewing box in a dimly lit room. Assessing the radiograph should be done in a step-by-step way.

The following procedure can be used when checking the films.

(1) **Positioning:** is the part rotated, is it the right area?
(2) **Centring:** is the primary beam centred on the area of interest?
(3) **Collimation:** is it 100%, 75%, 50%, 25% or 0%? This refers to whether the edges of the collimated beam are all visible. 75% collimation refers to one edge not being visible. From a safety point, all edges should be visible on the finished radiograph.
(4) **Exposure:** is it under- or overexposed?
(5) **Processing:** is it under- or overdeveloped, are there any chemical splashes?
(6) **Labelling:** is there a left/right marker, date and patient details?
(7) **Extraneous marks:** are there any white specks, finger-prints or scratches?
(8) **Overall assessment:** 'this is a great film of excellent diagnostic quality' or 'this is a poor film of little diagnostic value, better go and do it again!'

FILM FAULTS

General rules

It is important, firstly, to know the correct methods of setting exposures and processing film and, secondly, to recognise film faults and know how to correct them (Table 30.1).

Table 30.1 Some common film faults.

Fault	Cause
Too dark	Overexposure (reduce kV) Overdevelopment Developer temperature too high Excessive fog
Too pale	Underexposure Developing time too short Developing temperature too low Developer exhausted Developer too dilute
Too high contrast	kV too low
Too low contrast	kV too high Underdeveloped Excessive fog
Poor detail	Penumbra effect Short FFD Object–film distance too great Uneven screen–film contact Patient movement Grid lines No grid
Fog	Bad storage Chemicals Radiation Light (wrong filter)
Stained films – yellow (dichroic fog)	Insufficient rinsing Exhausted fixer
Streaking	Lack of agitation Dirty processing hangers Insuffcient rinsing Drying (water marks)
Crimp marks – crescent shape	Mishandling: Before exposure – black or white Before development – black
Abrasion marks	Mishandling Dirty rollers
Static marks – black streaks (look like lightening streaks)	Bad handling techniques
Developer splashes – black spots	Poor developing technique
Fixer splashes – white spots	Poor developing technique
Screen marks – white	Dust specks on screen
Finger marks – dark or light fingerprints	Poor film-handling technique

Dark films

Remember that overly dark radiographs are overexposed or overdeveloped (think of them as being burnt if it helps!). This is all very well, but how is it possible to tell if it is over*exposed* or over*developed*? The metal left/right marker will reveal the answer. Even if the film is overexposed, the metal marker should still show up white. If it has been overdeveloped, even the metal marker will appear darker.

Pale films

What about pale films? Easy, they are underexposed or underdeveloped! It is possible to tell the difference here because even on underexposed films, the exposed areas (around the body) should still be black because even the smallest exposure of X-ray photons on a film will cause blackening. The fault can be identified using the 'finger test'. Place a finger between the film and the light viewer in an exposed area that does not contain any part of the animal's body. If it is possible to see the finger through the film, the radiograph is underdeveloped. The most common cause of pale films is due to underdevelopment because the processing chemicals are old and exhausted.

Blurred image

The most common cause of blurring of the image is because the animal was moving at the time of exposure. It occurs most commonly because of respiratory movements and sometimes due to inadequate restraint of the patient. One should always try to keep the time of exposure as short as possible. The penumbra effect also causes blurring and distortion of the image. This occurs either due to a reduced film–focal distance (FFD) or long object–film distance.

Extraneous marks

Extraneous marks are white specks, scratches, etc. on the finished film. Bad screen handling and maintenance are the most common cause of this. Screens should be cleaned regularly and care must be taken when loading and unloading films from the cassette.

Chapter 31

Contrast Studies and other Imaging Techniques

Contrast studies may be required to gain additional information about structures that are radiolucent or are masked by adjacent or overlying structures. Contrast studies are useful for obtaining information about hollow organs, such as the size, shape and position of them. The VN must be aware of the different types of contrast available, their suitability for particular procedures and the patient preparation and administration of them. There are two methods of providing contrast:

- positive contrast;
- negative contrast.

POSITIVE CONTRAST RADIOGRAPHY

Positive contrast solutions are *radio-opaque* and so appear *white* on the finished film. They work because they consist of elements with high atomic numbers and they absorb a large proportion of the X-ray beam.

Two types of positive contrast preparations are available:

- barium sulphate;
- water-soluble iodine-based preparations.

Barium sulphate

This is a white, chalky substance, which is available as liquid, paste or powder. It is used exclusively for gastrointestinal tract studies and it provides excellent mucosal detail. Barium preparations are usually quite tasty and can be administered by mouth, either directly by syringe or stomach tube or mixed in with food. Barium preparations can cause pneumonia if aspirated and should not be used if a perforation along the gastrointestinal system is suspected as they can cause adhesions and granulomas.

Water-soluble iodine-based preparations

Ionic agents

Ionic iodine-based agents are water soluble and so they may be safely injected into the blood vessels. They may be used in a wide range of situations.

- *Intravenous urography.* These agents are excreted rapidly by the kidneys and so are used for intravenous urography (IVU) where they provide excellent outline of the upper urinary tract.
- *Cystography and urethrography.* These agents are also frequently used to perform retrograde urethrography or cystography, by injection of the contrast via a urinary catheter into the lower urinary tract.
- *Angiography.* The ionic agent is injected intravenously to demonstrate certain areas of the vascular system.
- *Arthrography.* The ionic agent is injected into joints to show up abnormalities in joint spaces.

Non-ionic iodine preparations

Non-ionic iodine-based solutions may be used for urinary studies etc., but they are very much more expensive and are mainly used for myelography as they do not irritate the spinal cord. These preparations are classified according to the concentration of iodine in mg/ml. For example Conray 420 contains 420 mg iodine/ml.

NEGATIVE CONTRAST AGENTS

Negative contrast appears *black* on the finished radiograph because it is radiolucent. It consists of gases – room air, oxygen, carbon dioxide or nitrous oxide. Negative contrast studies are useful for showing the position of organs, tumours within organs and urinary calculi. Room air is most frequently used and pneumocystograms are the most common procedure carried out using this method of contrast.

DOUBLE CONTRAST STUDIES

This contrast study uses both positive and negative contrast agents to enable good visualisation of mucosal detail of organs without obliterating small foreign bodies. A small amount of a positive contrast agent is administered into an organ, lining its internal surface, followed by a larger amount of air.

CONTRAST RADIOGRAPHY TECHNIQUES

Patient preparation prior to contrast studies

(1) Withhold food for at least 24 hours prior to barium studies and intravenous urograms.
(2) Administer enema prior to colonography, IVU, cystography and urethrography (because faeces will obscure abdominal features).
(3) Obtain plain views of the affected area first to check for any previously overlooked pathology and for comparison with the contrast radiograph.

(4) Warm the contrast medium first by placing the bottle in a kidney dish of warm water. The required amount can then be drawn up into a syringe and kept warm by holding it in one hand.

Gastrointestinal tract

- A barium swallow is used to evaluate the oesophagus and/or stomach (gastrogram) by giving the contrast medium by mouth.
- If a megaoesophagus is suspected the liquid barium may be added to tinned meat for the patient to eat (barium meal).
- The stomach may be evaluated by a positive contrast gastrogram. The barium may be administered by syringe or stomach tube. Better mucosal detail is seen using a double contrast gastrogram. The liquid barium is followed by introduction of room air.
- The small intestine can be assessed by performing a 'barium series', in which radiographs are taken at intervals after the barium administration.
- The large intestine may be studied in the following ways:
  - negative contrast study using air only (pneumocolon);
  - positive contrast study using a barium enema;
  - double contrast study using barium and air.

Urogenital tract

Intravenous urography (IVU) can be used to evaluate the kidneys and ureters. A water-soluble iodine-based contrast agent (e.g. Conray or Urografin) is injected intravenously and is rapidly excreted via the kidneys. Radiographs are taken in a series to highlight the kidneys and the ureters.

Contrast cystography can be used to study the bladder in the following ways.

(1) Negative contrast cystography (pneumocystogram) using air injected into the bladder via a urinary catheter.
(2) Positive contrast cystography using water soluble iodine contrast medium via urinary catheter.
(3) Double contrast cystography using (1) and (2) in combination.

The urethra can be studied by retrograde urethrography in male animals and retrograde vagino-urethrography in females. The urinary catheter is inserted into the urethra but pushed no further than the very caudal end of the urethra

The spine

Spinal disease can be investigated by myelography. Contrast medium is injected into the subarachnoid space which surrounds the spinal cord and contains cerebrospinal fluid (CSF). Special water-soluble non-ionic iodine preparations are used (Iohexol, Omnipaque) to minimise irritation. There are two approaches used to inject the contrast – cisternal puncture and lumbar puncture.

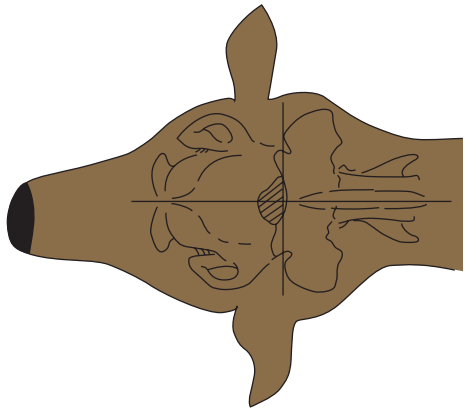


Fig. 31.1 The subarachnoid space is accessed at the position of the cross.

Procedure – cisternal puncture

(1) The animal is anaesthetised and plain radiographs of the spine are taken.
(2) The correct amount of contrast medium is drawn up (0.3 ml/kg).
(3) An area centred over the cisterna magna is clipped and surgically prepared.
(4) The animal is placed in lateral recumbency with the spine close to the edge of the table and the table tilted slightly to elevate the head.
(5) The neck is flexed ensuring the nose is parallel to the table. This helps to open up the gap between the skull and the atlas vertebra. Flexing of the neck will cause the endotracheal tube to kink, therefore a reinforced or armoured tube should be used or the tube can be temporarily removed during this procedure.
(6) Monitor the animal's respiration to ensure adequate ventilation at all times. The injection of the contrast sometimes causes temporary apnoea.
(7) The veterinary surgeon will then insert a spinal needle into the subarachnoid space (Fig. 31.1). After withdrawal of the stylet, CSF will drip from the hub of the spinal needle (Fig. 31.2). The contrast agent is then injected slowly. During this procedure it is essential that the head is kept in the same position throughout or the spinal cord may be severed.
(8) After injection of the contrast medium the needle is removed and the neck is extended. The table may be tilted more to encourage the flow of the contrast medium.
(9) Radiographs are then taken of the spine.

Procedure – lumbar puncture

If the lesion is more caudal then a lumbar myelogram is often performed.

(1) The animal is usually placed in sternal recumbency for preparation of the site.
(2) An area approximately 8 cm × 8 cm is clipped over the 3rd to 5th lumbar vertebrae. Injection is either between L3–4 or L4–5 vertebrae.

Fig. 31.2 Collecting CSF.

(3) The area is cleaned surgically and the animal positioned laterally with hindlegs pulled cranially or in sternal recumbency with the hindlegs again pulled cranially. The positioning helps to open the gap between the vertebrae.
(4) The veterinary surgeon then places a spinal needle into the subarachnoid space. Often little cerebrospinal fluid is seen but the animal's hindlegs may 'twitch', indicating correct position of the needle.
(5) The contrast medium is injected as before and several radiographs are taken.

After myelography it is important that the animal recovers with its head elevated. If a lot of contrast medium is allowed to flow into the brain there is a risk of seizures.

Heart

Angiocardiography under general anaesthesia may help in the diagnosis of some heart and vascular disease.

- Non-selective angiocardiography involves injection of a bolus of contrast via a catheter in the cephalic or jugular vein.
- Selective angiocardiography involves administration of contrast directly into the required heart chamber.

Portal venography

Certain liver problems can be diagnosed by portal venography. A laparotomy is performed under general anaesthesia. A hepatic vein is catheterised and

the contrast injected. Radiographs show the passage of the hepatic portal vein. Fluoroscopy is most commonly used for this procedure.

Arthrography

This is performed under general anaesthesia. Contrast is injected into the relevant joint via a needle and syringe. Strict asepsis is required when preparing the site so that bacteria are not introduced.

Fistulography

Contrast can be introduced into a sinus tract for its evaluation.

OVERVIEW OF OTHER IMAGING TECHNIQUES

Radiography is the most common method of obtaining diagnostic images of the body, however, over the past 20 years or so, other imaging methods have been developed and are used widely in the human field of diagnostics. These techniques are becoming more widely available to veterinary practices and include:

- ultrasound;
- computed tomography (CT);
- magnetic resonance imaging (MRI).

These other imaging methods will by no means replace the use and requirements for radiography, but they do provide their own specialised uses for certain diagnostic procedures.

Ultrasonography

Ultrasonography can be used to complement radiography and is particularly useful for:

- making organ measurements, e.g. kidney length, cardiac chambers;
- obtaining anatomical information not readily obtainable by other means, e.g. about the heart valves, biliary tract, pancreas and uterus;
- determining the origin and composition of masses, e.g. in the pleural and peritoneal cavities;
- investigating fluid accumulations;
- guiding percutaneous needle biopsy.

Ultrasound is sound at a frequency above the audible range, i.e. >20 000 Hz. Medical diagnostic ultrasound machines use frequencies in the range of 2–10 million Hz or 2–10 MHz.

The pulse–echo principle

Ultrasonography is based on the pulse–echo principle: by measuring the time taken for a pulse of sound to return to its source, the distance to the reflecting

structure producing the echo can be calculated. On a percentage basis, an ultrasound transducer emits sound <0.1% of the time and listens >99.9% of the time. A typical ultrasound pulse lasts a few milliseconds.

The ultrasound transducer is both the source of the sound and the receiver for echoes returning from tissues. These functions are possible using crystals with a property known as the *piezoelectric effect*. Applying a voltage causes the crystal to rapidly change shape and emit a sound. Conversely, incoming sound causes a slight deformity of the crystal, which produces a voltage. Naturally occurring piezoelectric crystals include quartz. The crystals in ultrasound transducers are of ceramic compounds. The size of the crystal determines its frequency; its shape influences the shape and direction of the emitted ultrasound beam. Specific crystal shapes have been designed that allow the beam to be focused.

The ultrasound image

There are three basic types of ultrasound image.

- **A-mode** is the simplest form of display, consisting of a series of spikes, which correspond to tissue interfaces. It measures two parameters: distance of the interface from the transducer and the amplitude of the echoes produced (A = amplitude).
- **M-mode.** Here the spikes are replaced by dots, the brightness of which is proportional to the amplitude of the echoes. The dots are displayed on a moving strip of paper or sweeping across a monitor screen producing a series of wavy lines from which measurements can be made. M-mode is used for making measurements of moving structures and to evaluate rapid motion of cardiac structures (M = motion).
- **B-mode.** This is also based on a line of dots of varying brightness (B = brightness). By collecting many diverging lines of dots, the familiar pie shaped, sector B-mode ultrasound image is produced. If a series of parallel lines of dots is collected the image is a linear scan. By collecting these lines of dots very rapidly a 'real-time' display is possible that shows the internal organs moving at the same time that they are actually moving in the body. Collection of many lines of ultrasound data and processing them into a real-time image requires very rapid computing.

The ultrasound beam is produced by the scanner probe (transducer) which contains special crystals, which vibrate when an electrical current (voltage) is applied across them. The voltage is applied in a pulsatile way and so pulses of sound are emitted from the transducer. When this transducer is then placed in contact with the body surface, these pulses of sound are passed through the body tissues. The different body tissues have different resistance to the passing sound, and each tissue type reflects or echoes the sound waves back towards the transducer in a different way.

The ultrasound machine converts these echoes in the transducer into small dots depending on their strength and depth of reflection. This collection of dots is built up into a picture and is shown on the screen. This gives a cross-sectional, moving image of the body area.

The resulting image is totally different to a radiograph. Dense soft tissues cause many echoes, which appear bright on the image. Less dense tissues produce fewer echoes and these give darker images. Fluids (e.g. urine) appear black on the image because they produce no echoes. On a radiograph, an organ such as the heart appears as one greyish-white mass. Ultrasound on the other hand, can illustrate the internal arrangement of the chambers, valves, vessels and myocardial thickness. Many cardiac anomalies can be identified by this method and accurate diagnoses made which then enable the most appropriate treatment to be initiated.

Doppler

In addition to measuring the amplitude of echoes returning from tissue, it is possible to measure their frequency. This is useful because moving structures (e.g. red blood cells) reflect ultrasound with an altered frequency. For example, an object moving towards the transducer will produce an echo with a higher frequency than was transmitted (frequency shift up), whereas an object moving away from the transducer will produce an echo with a lower frequency than was transmitted (frequency shift down). It is possible to measure accurately frequency shifts in the body and so calculate the velocity (direction and speed) of moving structures.

Veterinary applications of Doppler ultrasonography all involve the assessment of blood flow, such as:

- detection of altered intracardiac blood flow;
- detection of thrombosis;
- detection of portal hypertension.

Computed tomography

CT is radiological imaging technique that produces images of 'slices' through a patient's body. CT produces films of much higher quality and gives much finer tissue detail than that of conventional radiographs.

Based on the same principal as an X-ray machine, the patient is placed inside the CT scanner, whilst lying on a horizontal, moving bed. The X-ray tube head rotates at high speed around the chamber during the exposure and a computer processing system generates an image of the tissue density in a slice about 1 cm thick. This produces a three-dimensional picture of the anatomy which is the main benefit of CT scanning.

Some specialist referral veterinary clinics have an arrangement with their local hospital that enables them to use the scanner out of hours at arranged costs.

The main disadvantages of CT scanning methods are that the equipment is extremely expensive and maintenance costs are high. The ionising radiation risks associated with CT are much higher than with conventional radiography so no personnel can remain in the room during exposures.

Magnetic resonance imaging

MRI scanners produce similar cross-sectional views of the body as the CT scanner. Like the CT scanner, the patient lies within the chamber during the imaging process. MRI uses a combination of magnetism and radio waves to produce the image. It does this by analysing certain atomic elements in the body. During the scanning process, a series of radio waves are transmitted into the patient. The strong magnetic field and transmitted radio waves cause the atoms to vibrate in a certain way (this is called resonance) and they transmit a small radio signal. This signal is picked up by the scanner and converted into an image by a computer. Unlike CT scans, the cross-sections of the body can be obtained in any direction and the image produced provides exceptionally high detail to enable minute changes in the body tissues to be detected. This imaging technique is most widely used for studies of the brain.

The main disadvantage of the MRI scanner is that it takes longer (may take over an hour) and the machinery is hugely expensive to purchase, maintain and run. MRI scanning is safe, however. There is no scientific evidence to suggest that MRI is hazardous to personnel or patient. The main safety consideration is from the powerful magnetic field surrounding the scanner, which means that metal objects cannot be taken into the room of the scanner.

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SECTION 7

DIAGNOSTIC TESTS

Chapter 32

Introduction – The Laboratory

The VN should not only be familiar with running samples on modern biochemical and haematological machines but also be able to carry out simple but vital procedures in the laboratory. This section describes these simple procedures for easy reference.

HEALTH AND SAFETY

There are many potential hazards in the laboratory. It is essential that protective clothing is worn and that saftey rules are followed when carrying out procedures in the laboratory.

- Gloves and apron should always be worn when dealing with samples.
- Protective goggles should be worn when using the Bunsen burner.
- *Never* eat or drink in the laboratory.
- Clinical waste should be disposed of correctly.
- A wash basin should be available, preferably near to the exit.
- A first aid kit and eye wash should be easily located.
- Spillage kits should be available.

USING THE MICROSCOPE

The VN should be familiar with all laboratory equipment; correct usage and maintenance ensures accurate results. Fig. 32.1 describes the procedure for preparing the microscope for use.

Use a higher power objective lens for higher magnification.

Using immersion oil

When examining smears, immersion oil may be used. Place a drop of oil onto the smear and select the oil immersion lens. Slowly bring the lens down so that it is in the oil. Adjust the focus as necessary. Remember to clean the lens after use.

Vernier scale

The Vernier scale is necessary to relocate an object on the slide. By recording the two measurements, the object can easily be pinpointed again as long as

the slide is placed the correct way round on the stage. Each scale consists of a main scale with millimetre divisions and a smaller Vernier scale reading from 0 to 10 (Fig. 32.1k).

(1) The first part of the reading is taken from the main scale. Read the number opposite where the zero on the Vernier scale lies. If it is between two numbers, record the lower number.
(2) Then look to see which line on the Vernier scale lies directly opposite a line on the main scale. Take the reading off the Vernier scale.
(3) Repeat for the other scale. The two coordinates will be able to exactly locate a point on the slide in much the same way as grid references are used in map reading.

Fig. 32.1 Setting up and using the microscope.

MICROSCOPE AND SLIDE SET UP

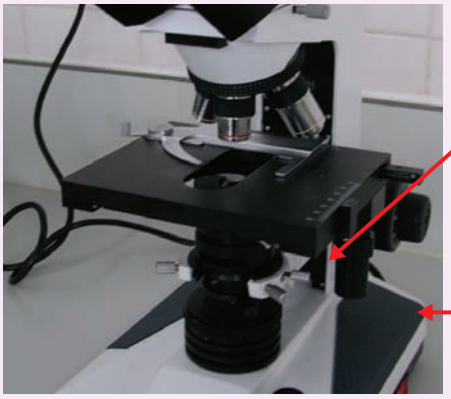
Check microscope is plugged in and switched on at the mains supply (Fig. 32.1a)

32.1a

Ensure that the (rheostat) light intensity switch is turned to the lowest setting (to avoid damaging the bulb and to extend the life of the bulb) (Fig. 32.1b)

32.1b

Turn the microscope on



Rack down the mechanical stage as low as possible (Fig. 32.1c)

using the coarse focus dial

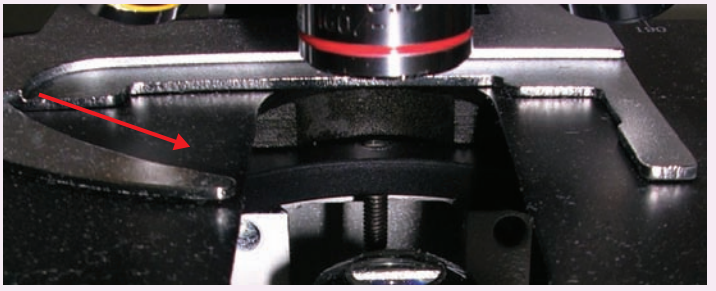
32.1c

Select the lowest power objective lens (×4 or ×10 depending on machine) and click into position (Fig. 32.1d)

32.1d

Place the microscope slide on to the stage (ensuring it is the right way up with the label to the right)

Ensure it is clipped into position using the slide retainers (Fig. 32.1e)



32.1e

Look at the stage directly, rack it up using the coarse focus so that the slide is positioned just below the objective lens

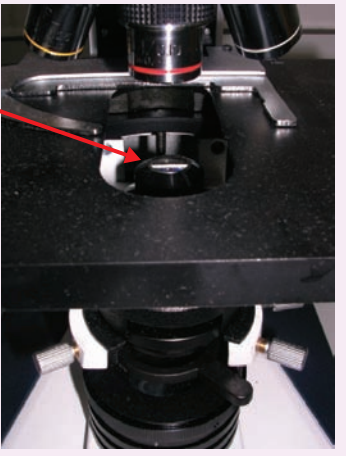
Continued

Adjust the rheostat as necessary (usually to a medium light intensity)

32.1f

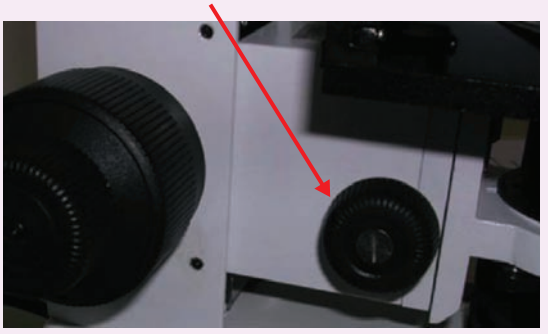
Adjust the binocular eyepieces to fit your eyes and to ensure that only one image is seen (Fig. 32.1f)

Check the position of the substage condenser and adjust to give the best possible image (usually racked up to just a few millimetres below the stage) (Fig. 32.1g)



32.1g

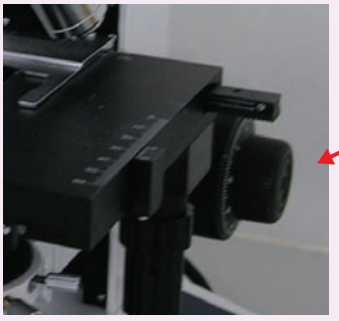
Substage condenser dial (Fig. 32.1h)



32.1h

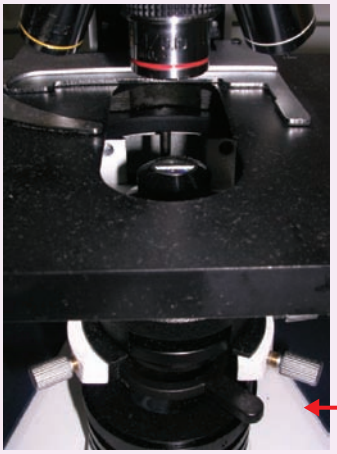
Move the slide using the position adjustment knobs so that the edge of the cover slip is under the lens

Rack the stage down using the coarse focus to focus the slide



32.1i

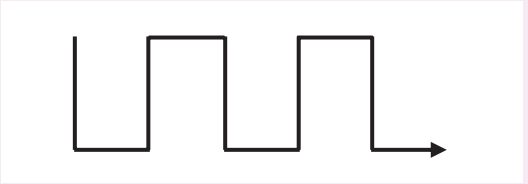
Adjust the fine focus (Fig. 32.1i)



Adjust the iris diaphragm to provide optimal lighting (Fig. 32.1j)

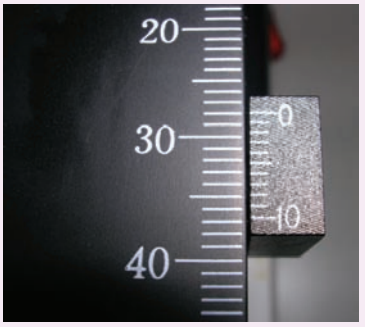
32.1j

Now use a higher power objective lens for higher magnification



Scan the slide methodically to locate the object of interest

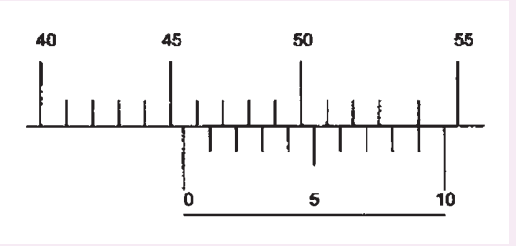
Once object located, position in the middle of the field and adjust fine focus if necessary



Reading the Vernier scale:
Read the horizontal scale first and then the vertical scale to obtain two readings which pinpoint an exact place on the slide (Fig. 32.1k)

32.1k

Continued



32.1l

Read off the larger main (Vernier) scale and note where the '0' of the smaller scale lines up (Fig. 32.1l)

If the '0' lines up in between two numbers, take the lower number (in the diagram this would be 45)

For the decimal point, read were the lines of both scales match up exactly and use the number from the small scale (in the diagram this would be 9)

Repeat the procedure for the vertical scale

Write down your reading

Chapter 33

Collection of Samples

BLOOD

Blood will normally clot after a couple of minutes outside the body. This may be necessary for certain tests where serum instead of plasma is required. Otherwise, an anticoagulant must be added to prevent coagulation. Manufactured blood tubes may be bought which contain anticoagulants. These blood tubes are colour coded and have push or screw on caps. Vacutainers have a different colour code from these tubes. Generally anticoagulants work by removing calcium, which is necessary for coagulation. However, heparin interferes with the formation and activation of thrombin. Table 33.1 compares blood tubes and their uses.

Blood sampling

In order to preserve the cells the blood should be collected carefully.

The VN should make sure everything ready before starting:

- swab (with skin preparation solution);
- syringes (appropriate size for patient – avoid excessive suction on the vein);
- needles (21–23 gauge);
- blood tubes;
- clippers/scissors;
- cotton wool swab to place on venepuncture site (tape also if necessary).

Site of collection

- Jugular vein (large vessel reduces the potential for cell damage).
- Cephalic vein.
- Saphenous vein.
- Femoral vein (in cats).

Preparation of site and method

- Clip hair away carefully.
- Clean and disinfect with chlorhexidine and then alcohol.
- After venepuncture, apply firm pressure on area.
- Remove needle from the hub of syringe before transferring to blood tube.

Table 33.1 Blood tubes.

Blood tubes	Vacutainer	Anticoagulant	Sample	Tests
Plain (white top) Gel tube (brown top)	Red Dark blue	None	Whole blood Serum	Biochemistry Serology
Pink	Lilac	EDTA	Whole blood	Haematology
Orange	Green or green/ orange	Lithium heparin	Whole blood	Biochemistry Electrolytes
Yellow	Grey	Fluoride oxalate	Whole blood	Glucose
Purple		Sodium citrate	Whole blood	Coagulation tests

Transferring the blood to the tubes

This needs to be done immediately if it is necessary to avoid coagulation.

- Remove needle from syringe before transferring blood into tube.
- Do not overfill the tube; only fill to the level marked on the tube.
- Replace the lid then gently invert/roll the tube to mix the blood with the anticoagulant, do not shake.
- Label the tube with owner's name, animal identification and date.

When filling a number of different tubes it is often routine for the EDTA tube to be filled first to avoid coagulation of the sample. However if a plain or gel tube is required, this should be the first tube to be filled as contamination with EDTA anticoagulant can cause abnormal potassium, calcium and magnesium results.

Notes on collection

- Insufficient anticoagulant will result in the sample clotting. Always ensure the blood tubes are filled accurately.
- Haemolysis (breaking up of red blood cells releasing haemoglobin) can easily occur, which may affect the results:
  - abnormally low red blood cell (RBC) count and low packed cell volume (PCV) (due to the destruction of RBCs);
  - interferes with the photometric biochemical estimations because of the strong red colour of the plasma;
  - abnormally high total protein level when measurements are taken using a refractometer.
- Ensure the animal has been starved for at least 8–12 hours before taking the sample to prevent collection of a lipaemic sample. The lipids in the blood give it a cloudy white appearance and this will also affect the results:
  - causes falsely elevated biochemistry values, because of the effect of the resultant turbidity of the sample on light transmission in photometric measurements.

Table 33.2 Urine preservatives.

Preservative		Analysis
Toluene (care – toxic)	Add enough so that a thin film forms on top of the urine	Biochemistry
Formalin (care – toxic)	Add 1 drop of 10% formalin to 2.5 ml urine	Good for examining urinary sediment, but interferes with biochemical tests. Kills bacteria
Thymol	Add 1 mg/ml of urine. Will preserve up to 24 hours	Biochemistry except glucose estimations. Kills bacteria
Boric acid	Add 0.5 g per 28 ml of urine. Sterile universal containers with red tops contain boric acid. Preserves bacteria	Bacteriology and sediment examination. Preserves the bacteria for up to 4 days

URINE

Time between collection and examination should be kept to a minimum. Once a urine sample has been obtained it should:

- be examined within 1 hour;
- be refrigerated and examined within 24 hours;
- be preserved and examined within 2 days.

Methods of collection

- Free-catch of mid-stream urine. If collected carefully, may be the most uncontaminated.
- Catheterisation. Always use sterile catheter. Some urethral bacteria may get pushed upwards.
- Cystocentesis. Clip and disinfect caudal abdomen area. Must be able to palpate the bladder first. Often carried out with ultrasound guidance.

Notes on collection

- Collect into a sterile container.
- Jam jars or ice cream pots, etc., should not be used for urine collection as these may affect the results.
- Ensure sterility at all times especially when collecting a sample for bacteriology.

A list of preservatives is given in Table 33.2.

FAECES

Examination of the faeces must take place as soon as the sample is collected to minimise changes, e.g. eggs hatching, dehydration. Faecal samples should

Table 33.3 Common test protocols.

Test	Procedure
Activated clotting time (ACT)	1) Preheat block and tubes (containing Fullers earth) to 37°C 2) Take 2 ml of blood into one of the tubes and mix well 3) Place back into the block. Start timing from moment blood sample was taken 4) After 1 minute examine sample for clotting and place back in the block 5) Then check sample every 10 seconds until blood starts to clot 6) Record when the sample first starts to clot
Adrenocorticotrophic hormone (ACTH) stimulation test	A blood sample is taken first (baseline) and then the tetracosactin (Synacthen™) is given intraveneously immediately afterwards The next sample is taken at least 30 minutes after the Synacthen™ was given. Synacthen™ should be given by intravenous injection at a dose of half a vial for dogs <15 kg and 1 vial for dogs >15 kg. The blood should be centrifuged and the plasma sent to the laboratory
Bile acids	Bile acids are run routinely in the biochemistry profiles. If a postprandial bile acid sample is needed, check that blood taken was from a fasted patient. If not take a preprandial sample first and then feed the animal (hand or force feed if necessary). A blood sample should then be taken 2 hours after feeding and placed into a heparin tube
Blood cultures	Take three samples each an hour apart. Care must be taken to use an aseptic technique, i.e. clip and prepare neck. Raise jugular vein and assess its position by sight, do not touch area with fingers. Take 10 ml of blood. Change needles and inject into blood culture bottle. Label bottle with animal's details including date, time, etc. Store bottles in the fridge prior to submission
Cortisol	Serum/plasma sample sent to laboratory
Digoxin levels	Serum sample sent to laboratory
Feline leukaemia virus (FeLV), feline immunodeficiency virus (FIV)	For FeLV and FIV send either serum or plasma
Feline infectious peritonitis (FIP)	Serum samples are needed
High dose dexamethasone suppression test (HDDST)	Blood samples should be collected into heparin tubes at 0 hours, 3 hours and 8 hours postinjection. Dexamethasone should be injected at a dose of 0.1 mg/kg by intravenous injection *Note*: A soluble dexamethasone preparation such as Dexadreson™ (2 mg/ml) should be used
Low dose dexamethasone screening test (LDDST)	Blood samples need to be collected into heparin tubes at 0 hours, 3 hours and 8 hours postinjection. Dexamethasone should be injected intravenously at a dose of 0.01 mg/kg. The blood samples should be centrifuged and the plasma sent to the laboratory *Note*: A soluble dexamethasone preparation such as Dexadreson™ (2 mg/ml) should be used

Table 33.3 *Continued*

Test	Procedure
Taurine levels	Laboratories can run taurine along with 15 other amino acids using a chromatography method on serum
Total T4 (thyroxine)	Test is run on serum/plasma samples sent to laboratory
Thyroid hormone stimulation test	Collect blood samples into heparin tubes at 0 hours and 6 hours postinjection. Thyroid stimulating hormone (TSH) should be given by intravenous injection at a dose of 0.1 unit/kg (TSH contains 2 units/ml), up to a maximum dose of 5 units or 2.5 ml
Von Willebrand's disease	Blood should be put into a citrate tube for the laboratory

be put into an airtight container preferably filled to the top to prevent further deterioration. Formalin may be added for preservation.

LABORATORY TEST PROTOCOLS

There are some tests that are performed by outside laboratories. Each laboratory will have its own requirements on how samples are to be collected and sent. Table 33.3 lists some common tests; collection methods are usually the same for each laboratory. If in doubt, contact the specific laboratory.

Chapter 34

Blood Tests

Much can be determined from relatively small amounts of equipment in the practice laboratory. This increases the speed at which samples can be processed and cases treated.

HAEMATOLOGY

Haematology involves evaluating the quantitative measurement of the cells and also the qualitative assessment including changes in cell morphology.

Haematocrit or packed cell volume

This is used to find the percentage of the blood that is occupied by red blood cells (RBC). It is used to assess anaemia or dehydration. It can also be used to measure plasma protein levels and these two results should be interpreted together.

The method for measuring packed cell volume (PCV) is illustrated in Fig. 34.1. The percentage may also be read by measuring the length of the red blood cells and dividing it by the total length of the plasma, buffy coat and red cells. Multiply this by 100 to get the percentage.

Measuring plasma protein

Once the PCV measurement has been recorded, the microhaematocrit tube is scored with a diamond pen or razor blade just above the buffy coat. A drop of the plasma is then placed on the prism of the refractometer and the result read off the appropriate scale. Plasma protein may also be measured using a biochemical analyser. Measurement units are either in g/dl or g/l.

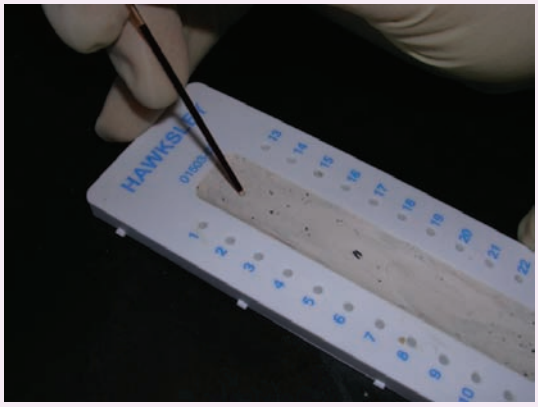
Blood smear

Blood smears are useful to examine cell morphology and for a differential white blood cell count. The smear should be produced soon after collection. Hold the spreader slide at a large angle for anaemic blood and a smaller angle for haemoconcentrated blood as this helps to concentrate or spread the cells of a blood smear. See Fig. 34.2.

It should be possible to assess the smear and diagnose any faults using Table 34.1.

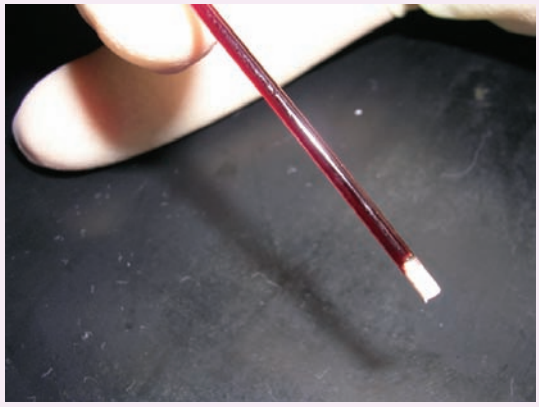
Fig. 34.1 Performing Packed Cell Volume test.

METHOD
Wear gloves
Select EDTA (pink) or Heparin (orange) tube (Not clotted or overfilled)
Mix sample gently
Remove two capillary (microhaematocrit) tubes from container
Remove lid from selected blood sample tube
Hold sample tube and capillary tube within the tube at a slight angle to enhance the flow of blood into the capillary tube
Fill capillary tube with blood *at least* ¾ full
Place a finger over one end of the tube
Wipe the outside of capillary tube with tissue
Plug one end of the tube with soft-clay sealant (Fig. 34.1a)



34.1a

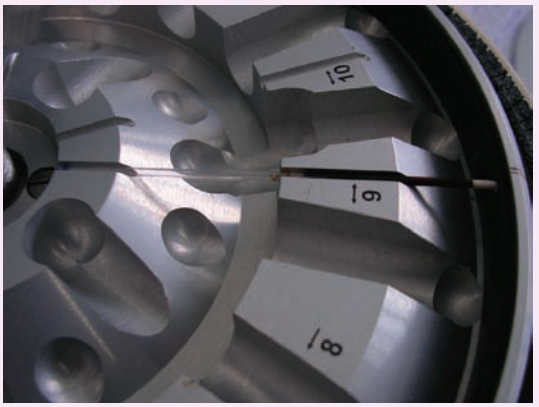
(Fig. 34.1b)



34.1b

Continued

Place tube into microhaematocrit centrifuge



Clay plug facing outwards against outer rim (Fig. 34.1c)

34.1c

Repeat the procedure to fill another capillary tube

Replace lid of blood sample tube

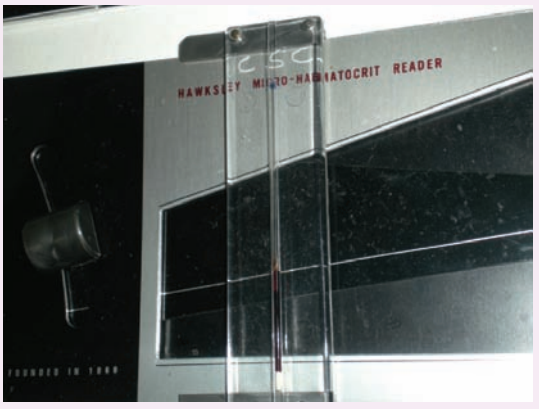
Place second tube into centrifuge (plug facing outwards) – opposite the first

Screw inner safety lid down over samples

Close *and* lock main lid

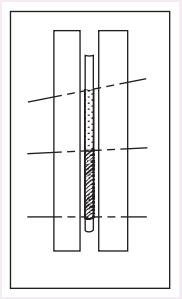
Set at 10,000 rpm for 5 minutes or 'fast' setting

Obtain PCV reading



34.1d

Place capillary tube in microhaematocrit reader. Correctly read prepared sample (Fig. 34.1d)



(Fig. 34.1e)

34.1e

Record the result

Fig. 34.2 Preparing a blood smear.

Wear gloves

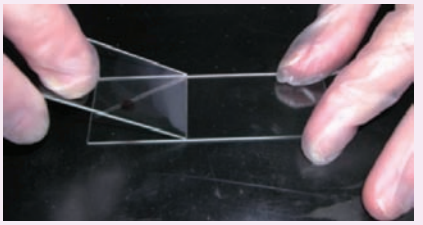
Select EDTA sample (not clotted or overfilled)

Rotate sample gently

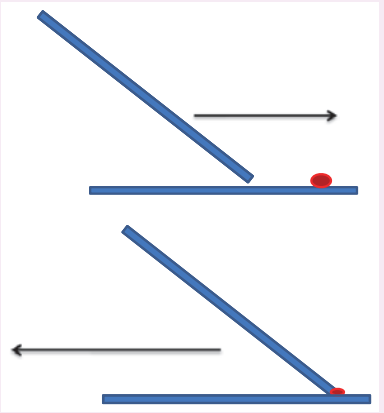
Use capillary tube, place small dot of blood near one end of the slide

Place spreader on main slide at the other end to the blood sample

Hold spreader at 30–45° angle (Fig. 34.2a)



34.2a



34.2b

Draw back spreader to contact blood, allow blood to spread (Fig. 34.2b)

Continued

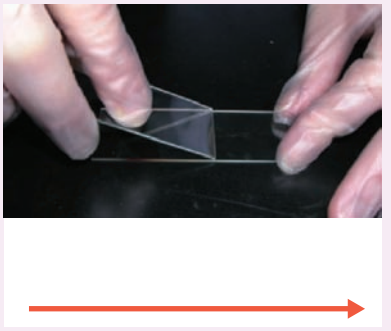
Push spreader in one smooth, rapid stroke along the microscope slide (Fig. 34.2c)	 34.2c
Air dry slide	
Label slide	

Table 34.1 Identification of faults on blood smears.

Fault	Cause
Too thick	Drop of blood too large
Too thin	• Drop of blood too small • Blood spread too quickly
Bands of thick and thin	'Hesitation bands' – caused by not spreading blood smoothly
Streaks along sample	• Spreader has chipped edge • Dried blood on edge of spreader • Dust on slide or in blood
Spots (blood absent)	If grease is present blood will not stick to the slide
Narrow thick smear	• Not waiting for blood to run along edge of spreader • Spreader not in complete contact with the slide
Crenation	Occurs if solution outside cell is hypertonic and fluid passes out of cell. Cells appear shrivelled with spikey edges
Incorrectly stained	• Smear not completely covered by stain • Distilled water at wrong pH (not 6.8) • Old stain (crystals form) • Stain not timed accurately • Over decolorised by washing slide too much • Insufficient mixing • Putting stains on in wrong order • Squirting with water too hard when washing

Staining blood smears

The blood smear is usually stained enabling better visualisation of the white blood cells which take up the stain. The most commonly used stains are from the Romanowsky group.

Leishman's staining method

Equipment:

- dish;
- rack;
- Leishman's stain;
- distilled water.

Method.

(1) Place slide on rack, smear uppermost.
(2) Cover with Leishman's stain. Leave 1–2 minutes to fix smear.
(3) Cover slide with *twice* volume of buffered distilled water (pH 6.8).
(4) Gently rock slide from side to side to mix solution evenly. Leave for 10–15 minutes.
(5) Wash solution off slide with the buffered distilled water. Flood slide for 1 minute until pinkish tinge just appears.
(6) Pour off the water and stand the slide upright on blotting paper to dry.

Giemsa stain

This is useful for identifying particular cells or parasites (e.g. *Haemobartonella felis*).

Method.

(1) Dip the slide in methanol for a few seconds to fix the cells.
(2) Flood the slide with Giemsa and leave for 30 minutes.
(3) Rinse the slide with distilled water and leave upright to dry.

Diff-Quik™ stain

This is a very quick and easy method of staining smears although not as good as Leishman's. The different dye solutions are combined in Leishman's stain and this gives rise to a coloured salt which is also involved in staining the cell components. This leads to improved staining of the smear. For Diff-Quik stain, three solutions are required:

- solution A – fixative (methanol) light blue;
- solution B – stain (eosin) red;
- solution C – stain (thiazine dye) purple.

Table 34.2 Normal ranges for differential white blood cell counts.

White blood cell	Dog %	Cat %
Neutrophils – adult	70	60
– immature	0.8	0.5
Eosinophils	4	5
Basophils	1	1
Lymphocytes	20	32
Monocytes	5	3

Method.

(1) Dip slide into solution A five times.
(2) Remove and dip into solution B five times.
(3) Remove and dip into solution C seven times (helps to stain the platelets if left longer).
(4) Rinse the slide with distilled water and leave upright to air dry.

Differential white cell count

This involves examination of the blood smear and calculation of the different types of white blood cell present.

Equipment needed:

- stained blood smear;
- immersion oil;
- microscope;
- chart to count cells.

Method.

(1) Prepare a fresh blood smear and stain with Leishman's or Diff-Quik™.
(2) Apply a small drop of immersion oil to the edge of the smear.
(3) Move along the edge of the smear using the 'battlement' method.
(4) Count and record the different types of leucocyte.
(5) Count at least 100 cells for every 10 000 cells in the total white cell count. The more cells that are counted the more accurate the result will be.
(6) Work out the percentage for each type of cell found. See Table 34.2.

Total cell counts

Total cell counts (complete blood count – CBC) can be performed manually using an Improved Newbauer haemocytometer and special dilution pipettes (Thoma). This technique has been superseded by automated cell counters.

Chapter 35

Urine Tests

Various simple tests can be performed on urine with minimal equipment:

- dipstick analysis: biochemical estimations of glucose, protein, ketones, bilirubin and pH, etc.;
- specific gravity: an index of the concentration of substances dissolved in it;
- examination of the sediment.

URINE DIPSTICK TESTS

There are a number of different types of dipstick available. Some have up to eight different chemical tests including nitrite, pH, protein, glucose, ketone bodies, urobilinogen, bilirubin and blood. Others are more specific, e.g. measuring just glucose.

The method is very similar for each test strip, however the following points are important to note in order to get reliable results.

- Ensure the strips are 'in date' and have been stored in their original container at the correct temperature.
- Follow the enclosed instructions carefully with regard to checking the colour change at the correct time.
- The urine should be fresh (<4 hours old).
- Do not 'dip' the stick in the urine. Instead use a pipette to remove a sample of urine and ensure the pads are all covered. This allows the urine sample to be used for other tests.

SPECIFIC GRAVITY

The specific gravity of urine is an estimation of the concentration ability of the kidney.

$$\text{Specific gravity} = \frac{\text{weight of a certain volume of a liquid}}{\text{weight of the same volume of water}}$$

Therefore the specific gravity of pure water is 1.000.

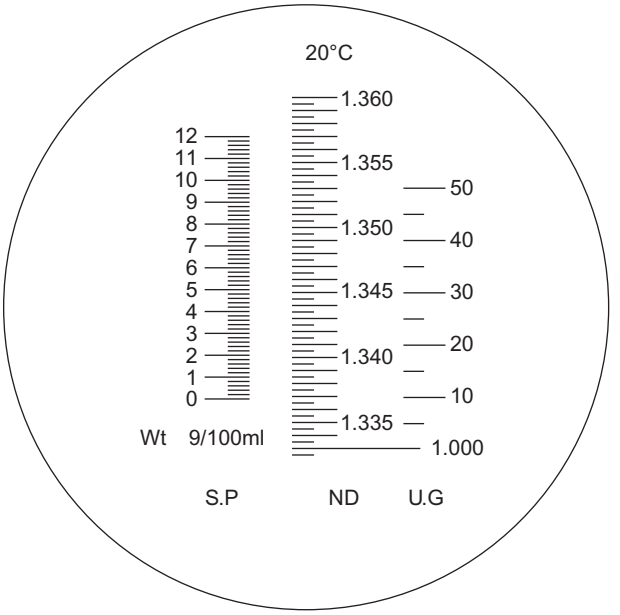


Fig. 35.1 Refractometer scale.

Measuring specific gravity using a refractometer

Equipment:

- urine sample;
- refractometer;
- distilled water;
- tissues;
- disposable gloves;
- pipette or 1 ml syringe.

Method.

(1) Put on disposable gloves.
(2) Test with distilled water – place 1–2 drops on the face of the prism and replace the cover.
(3) Look through the eye piece, pointing the refractometer towards the light. Focus scale by rotating rim of eyepiece. The line should cross at the 1.000 mark, if not alter using the scale adjusting screw (Fig. 35.1).
(4) Wipe away water with a piece of soft tissue.
(5) Then repeat procedure using 1–2 drops of urine.
(6) Record result.
(7) Clean refractometer using distilled water.

Note: The refractometer is calibrated for use at room temperature. Urine that has been stored in the fridge should be allowed to come to room temperature

before testing. If the urine is particularly concentrated it may give a result which is off the scale. In this case, dilute the urine with the same volume of water and take another reading. The results should then be doubled to get the actual reading.

The normal range in healthy animals is:

- dog: 1.015–1.045;
- cat: 1.035–1.060.

There may be some variation from this depending on fluid intake and hydration status.

EXAMINATION OF URINARY SEDIMENT

The urine sample may be centrifuged so that any solid particles are concentrated into sediment. A normal sample will not usually produce much sediment.

Preparation of the sediment

Equipment:

- fresh urine sample;
- centrifuge and centrifuge tubes;
- Sedistain™;
- pipette or 1 ml syringe;
- microscope slide and cover slip;
- disposable gloves.

Method.

(1) Centrifuge sample for 5 minutes at 2000 rev/min in a centrifuge or Eppendorf tube.
(2) Remove the supernatant fluid using a pipette, leaving a few drops in which to resuspend the sample. 'Flick' the tube to do this.
(3) A drop of Sedistain™ may be added in which to resuspend the sediment and to stain the debris.
(4) Place a few drops on a microscope slide and carefully apply a cover slip, avoiding air bubbles.
(5) Examine under low power and low illumination.

Producing a smear of the sediment

Equipment:

- fresh urine sample that has been centrifuged;
- Sedistain™;
- pipette or 1 ml syringe;

- microscope slide and 'spreader';
- disposable gloves.

Method.

(1) Prepare the sample as before.
(2) Place a small drop of the sediment on the slide and smear using the same method as for blood.
(3) Alternatively, use a bacteriological loop to spread the drop evenly over the slide.
(4) Allow the smear to air dry.

Staining the preparation:

- Leishman's stain will stain cells;
- Gram's stain will stain bacteria;
- Sedistain™ stains all of the sediment.

The sample may then be examined for crystals, casts, bacteria, cells and blood cells.

Chapter 36

Faecal Tests

Examination of the faeces must take place as soon as the sample is collected to minimise changes, e.g. eggs hatching, dehydration. Faecal examination is an important part of diagnosing gastrointestinal disease. The tests usually involve identification of parasites or bacteria. Trypsin digestion tests, involving a range of faecal solution dilutions and radiographic film, have now been replaced with more accurate blood tests for endocrine pancreatic insufficiency such as the trypsin-like immunoreactivity (TLI) assay.

PREPARATION OF A DIRECT FAECAL SMEAR

A faecal smear can provide evidence a range of different faecal protozoans such as *Giardia* and *Isospora*. Bacterial endospores may also be identified using appropriate stains. The faeces may be cultured on bacterial plates, however only colonic bacterial flora may be seen rather than small intestinal. Parasite eggs may be seen, however the concentration methods give better results. Various stains may be added to a faecal smear to identify undigested components of the animal's diet but these are often unreliable.

Equipment:

- fresh faecal sample;
- water or saline;
- microscope slide and cover slip;
- spatula;
- disposable gloves.

Method.

(1) Put on gloves.
(2) Place a few drops of water or saline on the microscope slide.
(3) Mix in a small amount of faeces (remove large pieces of debris).
(4) Apply a cover slip (avoiding air bubbles) and examine under low power.
(5) Examine the smear systematically, e.g. from left to right across the smear then move down one field of vision, then examine from right to left and so on.

CONCENTRATION METHODS FOR DETECTING ENDOPARASITIC OVA

Sedimentation

This method is useful as any ova present will be concentrated into the sediment.

Equipment:

- fresh faecal sample;
- glass container or jar;
- spatula;
- sieve;
- centrifuge and centrifuge tubes;
- disposable gloves and tissues.

Method.

(1) Put on gloves.
(2) Add 2 g faeces to 30 ml water in a small jar or bottle.
(3) Mix thoroughly to break up the faecal matter. Glass beads may be added and the solution shaken vigorously.
(4) Strain into a conical centrifuge tube to remove gross debris.
(5) Centrifuge for 3 minutes at 1–1500 rev/min.
(6) Discard the supernatant fluid and pipette a few drops of the sediment on to a microscope slide. Apply a cover slip avoiding air bubbles.
(7) Examine under low power.

SEDIMENTATION AND FLOTATION

This is the same technique as above but a flotation solution is used instead of water. After discarding the supernatant fluid, the tube is filled completely with a flotation solution. A cover slip is placed on top of the meniscus formed and left for 10 minutes. The eggs will float up to the surface. Thereafter the cover slip can then be transferred directly to the microscope slide.

Flotation solutions include:

- saturated sugar solution;
- saturated salt solution;
- sodium nitrate solution;
- zinc sulphate.

McMASTER TECHNIQUE FOR COUNTING WORM EGGS

This is a quantitative method for determining the number of eggs per gram of faeces. It is not routinely used, as any evidence of ova will be treated with appropriate anthelmintics.

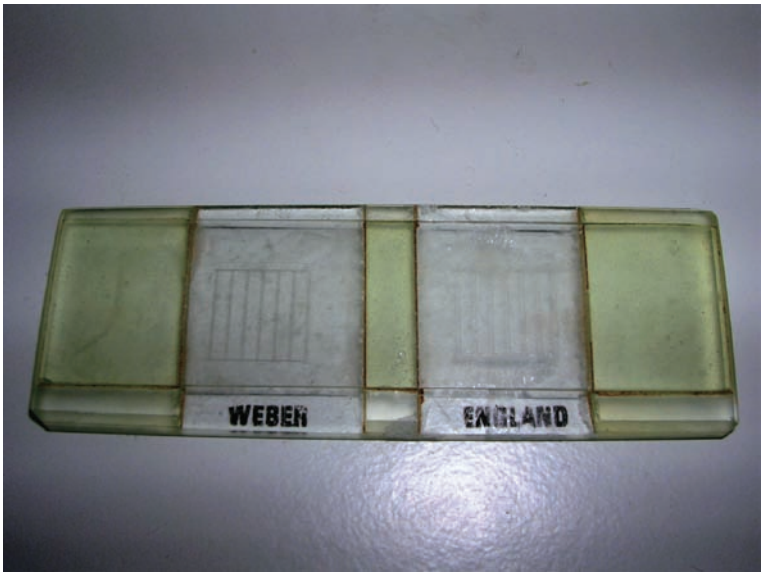


Fig. 36.1 McMaster slide.

Equipment:

- McMaster counting chamber (Fig. 36.1);
- fresh faecal sample;
- flotation solution;
- pipette or syringe;
- disposable gloves and tissues.

Method.

(1) Put on gloves.
(2) Mix 2 g of faeces with 30 ml of a flotation solution.
(3) Place in a screw-top jar (with glass balls).
(4) Shake well for 1–2 minutes.
(5) Strain, stir and pipette solution into both counting chambers.
(6) Examine under low power and count eggs in both chambers.

Calculation:

$$X = \text{number of eggs in both chambers.}$$

$$X/2 \times 100 = \text{number of eggs per gram of faeces.}$$

Chapter 37

Bacteriology Tests

Most bacterial tests are undertaken at commercial laboratories, mainly for safety and accuracy reasons. Smears can easily be performed in house, and stained for preliminary identification of bacteria.

MAKING A BACTERIAL SMEAR

Equipment:

- microscope slide;
- Bunsen burner;
- tongs for holding the slide when heat fixing;
- chinagraph pencil for labelling the slide;
- disposable gloves, face mask, goggles.

Method.

(1) Take a clean microscope slide and sterilise it by passing it through a Bunsen burner flame.
(2) Allow the slide to cool then apply sample material to the slide.
(3) Allow the smear to air dry.
(4) When the smear is dry, fix it by passing it through the flame to kill the bacteria and adhere them to the slide.
(5) Before staining use the chinagraph pencil to label the slide and to outline the smear.

STAINING BACTERIAL SMEARS

The bacterial smear can then be stained to identify different shapes or families of bacteria.

Methylene blue

This is a simple stain to show the shape of the bacteria.
Equipment:

- heat-fixed smear;
- methylene blue 1% solution;

- distilled water;
- staining rack;
- disposable gloves.

Method.

(1) Put on gloves.
(2) Place slide on the rack with smear uppermost.
(3) Flood slide with 1% aqueous methylene blue.
(4) Leave for 2 minutes.
(5) Wash off with distilled water.
(6) Leave slide upright to dry.

Gram stain

This stains Gram-negative and Gram-positive bacteria different colours in order to identify them.

(1) First stage: the *violet* (primary or basic stain) stain stains both types of bacteria.
(2) Second stage: the *mordant* (iodine) fixes the violet stain.
(3) Third stage: the *decoloriser* (alcohol) causes the Gram-negative bacteria to lose their violet colour.
(4) Fourth stage: the *red* stain (carbol fuchsin) stains the Gram-negative organisms red. The Gram-positive organisms remain violet.

Equipment:

- heat-fixed bacterial smear;
- crystal violet;
- Gram's or Lugol's iodine;
- acetone or industrial methylated spirit;
- dilute carbol fuchsin (1/10 with distilled water) or safranin 0.5%;
- staining rack;
- distilled water;
- disposable gloves.

Method.

(1) Put on gloves.
(2) Place the slide on the rack with the smear uppermost.
(3) Flood slide with crystal violet solution.
(4) Leave 30–60 seconds.
(5) Gently rinse the slide with distilled water.
(6) Flood with Gram's or Lugol's iodine.
(7) Leave 30–60 seconds.
(8) Pour off excess iodine and rinse with distilled water.
(9) Flood slide with acetone and immediately rinse off with distilled water.
(10) Counterstain with dilute carbol fuchsin for 1 minute.

(11) Pour off excess stain and rinse with distilled water.
(12) Leave slide upright to air dry.

Lugol's iodine is more concentrated than Gram's iodine (1 ml Lugol's added to 100 ml water = Gram's iodine). It is used for a darker colour, and there is less chance of excessive decolorisation.

Ziehl-Neelson acid-fast stain

This stain is used to detect 'acid-fast' organisms, e.g. *Mycobacterium tuberculosis*. The slide is stained with carbol fuchsin and heated. Acid-fast organisms are resistant to decolorisation with the acid alcohol and so retain the red colour when counterstained with methylene blue. Firstly all bacteria stain red. Then acid-alcohol washes red stain off non-acid-fast bacteria. These bacteria then stain red–blue.

CULTURING BACTERIA

Most bacteria can be grown on agar plates at 37°C. Small incubators are available that can be used in the practice. However, there are potential biosecurity risks and lack of expertise in identifying the bacteria which may lead to inaccurate diagnoses.

Inoculating agar culture plates ('streaking out' method)

The bacteria are spread out over the plate using the streaking out method. This allows single colony identification.

Equipment:

- agar plate (sterile Petri dish with appropriate agar growth medium);
- inoculating loop;
- Bunsen burner;
- bacterial sample;
- disposable gloves, face mask and goggles.

Method.

(1) Put on gloves, mask and goggles.
(2) Sterilise the inoculating loop in the Bunsen burner flame until it glows orange.
(3) Pick up the half of the Petri dish containing the media and turn to face upwards.
(4) Smear the sample from the loop or swab over a small area to produce a 'well' of bacteria (well inoculum) (Fig. 37.1).
(5) Resterilise the loop in the flame or replace the swab in its container.
(6) With the sterile loop make three to four short strokes all in the same direction away from the well (this will be A). Take care not to tear at or cut the agar.

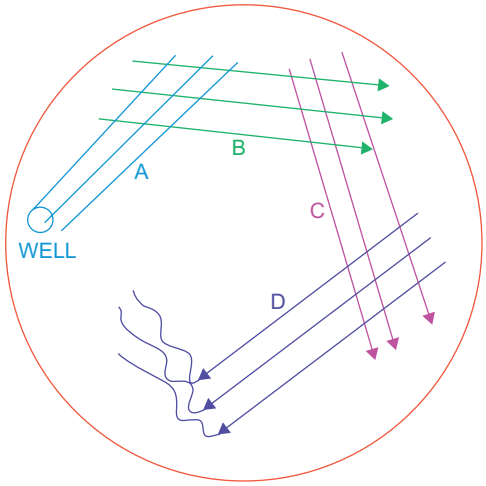


Fig. 37.1 An inoculating plate with streaking out method.

(7) Resterilise the loop.
(8) Continue in the same way to produce streaks B, C and D. Resterilise the loop between each group of streaks and then again at the end. In this way the bacteria are thinned out, so that by groups C and D, individual colonies of bacteria will grow.
(9) Place the two halves of the Petri dish together, the one containing the medium uppermost.
(10) Label with case details and incubate at 37°C for 18–24 hours. Plates must be laid in the incubator medium side uppermost to prevent condensation contaminating the sample.

Identification of bacterial growth may depend on colony characteristics:

- any zone of haemolysis in the blood agar around the colony (haemolytic or non-haemolytic);
- the size of the colony (measurement in millimetres);
- the colour of the colony (grey, cream, yellow, white, etc.);
- if opaque or translucent;
- the shape and consistency (irregular, circular, raised, flat, convex, mucoid, flaky, hard or crusty);
- the odour (sweet, musty, pungent, etc.).

Inoculating culture plates for use with sensitivity discs

The bacteria are initially cultured on the appropriate agar. Colonies may then be taken and spread over the sensitivity testing agar and antibiotic-impregnated discs applied. The plate is then incubated again.

Equipment:

- diagnostic sensitivity testing agar;
- antibiotic-impregnated sensitivity discs;
- inoculating loop.

Method 1.

(1) Make an inoculum well.
(2) Make strokes from the well in three directions across plate to spread sample as evenly as possible without sterilising the loop in between (sterilise only at the beginning and end).

Method 2.

(1) Take a sterile syringe case and add 1 ml of sterile water.
(2) Sterilise the loop and pick up the sample on the loop.
(3) Mix the loop full of sample in sterile water.
(4) Pour over the medium, then rock the dish to spread the sample evenly over the surface. After this, pour off the remaining solution.

After inoculating the media, the sensitivity discs can then be applied to the surface of the agar using a sterile pair of forceps or a purpose-designed disc dispenser. Sensitivity discs are paper discs impregnated with different antibacterial and antibiotic agents. The discs should be firmly pressed on to the agar surface using sterile forceps (sterilise the forceps by passing them through the Bunsen flame). This allows the antibacterial substance to diffuse into the surrounding agar. The plates are incubated at 37°C and checked after 18–24 hours. The agar surface should be covered with bacterial growth except for around discs that contain an antibiotic or antibacterial agent that those bacteria are sensitive to.

It is not completely accurate to record sensitivity in this way because some bacterial substances diffuse out into the media more quickly than others and the concentration of drugs on the discs may not compare to the normal doses given to the patient.

Chapter 38

Dermatology Tests

HOW TO FIND ECTOPARASITES

Collection of samples

Many parasitic skin diseases appear very similar to each other clinically, so sampling methods must be undertaken and laboratory analysis carried out for accurate diagnosis. The VN must be confident in obtaining samples and performing the various laboratory tests to aid diagnosis. Good sample quality is essential to get reliable results.

It is essential to take appropriate hygiene precautions, i.e. wear gloves, and an apron if necessary. The sample being taken may be zoonotic (e.g. *Sarcoptes*, *Cheyletiella*, ringworm).

The following methods may be performed to collect ectoparasite samples:

- skin scrapings;
- coat brushings;
- hair plucks;
- tape strips;
- pustular smears;
- ear wax swab.

Skin scrapings

Skin scrapes are useful to detect the burrowing mites *Sarcoptes scabei* and *Demodex canis*.

Method.

(1) Chose a suitable area (usually the edge of the affected area or lesion is better because the mites travel outwards from the centre).
(2) Gently clip hair around the area to be scraped.
(3) Apply a drop of liquid paraffin to the skin surface and a no. 10 scalpel blade.
(4) Squeeze the skin between the fingers so that the area is stable. This also helps to squeeze the parasites closer to the surface.
(5) Gently scrape the top layers of the skin surface until pin-pricks of blood are seen oozing from the area. It is important to scrape until blood appears to ensure that the scrape is deep enough.

(6) Transfer the scraped material on to a microscope slide and smear it out so that the sample is not too thick; this will make it easier to see the parasites.
(7) Place a cover slip over the top and examine the slide under low power.
(8) Scan the slide methodically until the parasite is found.
(9) Increase magnification as necessary.
(10) Record the Vernier scale readings of any mites found so that they can be relocated if necessary.

Some people prefer to place a few drops of 10% potassium hydroxide (KOH) on to the slide instead of liquid paraffin because it removes (clears) the skin cells and keratin. However, liquid paraffin is useful because the parasites remain alive, making location of them far easier. Take several scrapings from several sites, using a new blade for each site.

Coat brushings

Coat brushings may reveal fleas and/or their faeces, lice, *Cheyletiella* mites and eggs.

Method.

(1) Stand the patient on a large sheet of white paper.
(2) Vigorously brush the coat with the fingers or brush.
(3) Collect the dislodged material and transfer some of it on to a microscope slide with some liquid paraffin.
(4) Apply a cover slip.
(5) Examine under the microscope in the same way as a skin scraping.

Wet paper test

Wet a small wad of cotton wool and squeeze all of the excess moisture out of it. Dab this over the remaining sample that is left on the white paper from the coat brushing. Flea faeces may be confirmed by the blood-tinged marks on the cotton wool or paper.

Hair plucks

Hair plucks may be taken and submitted for fungal culture or examined for louse and *Cheyletiella* eggs, which are attached to the hair shaft.

Method.

(1) Pluck small amounts of hair from the affected area.
(2) Place on to microscope slide with some liquid paraffin if examining for eggs.
(3) Place cover slip and examine under low power.

When examining a hair pluck for the presence of fungal spores (for evidence of ringworm), place sample on to a microscope slide but do not add

any liquid paraffin or KOH. Place a cover slip and examine for spores under ×100 or ×400 magnification. It is also possible to examine these hairs under ultraviolet light (Wood's lamp) for fluorescence to confirm the presence of *Microsporum canis*.

Tape strips

Clear adhesive tape (e.g. *Sellotape*™) may be used to collect fleas, lice and harvest mites and to demonstrate the presence of microbes such as *Malassezia*.

Method.

(1) Choose the area to be tested and, if necessary, separate the hairs to allow access to the skin below.
(2) Press a length of clear adhesive tape firmly on to the area.
(3) Repeat this and then transfer the tape, sticky side down on to a microscope slide for examination under low power.
(4) If examining for *Malassezia*, loop the section of tape with the sample on and stick the remaining tape to a microscope slide.
(5) Dip the sample into a stain such as Diff-Quik™ or methylene blue and examine under oil immersion for presence of yeasts and fungal hyphae.
(6) Unstick the tape and reposition so that the sample is over the slide.
(7) Examine under high magnification.

Pustular smears

Pustular smears are useful to detect pustular demodectic mange and for identification of bacteria from skin lesions.

Method.

(1) Squeeze a pustule and transfer the contents directly onto a microscope slide (wear sterile gloves to hold skin for bacterial examination).
(2) Smear the contents evenly over the slide with a mounted needle or similar.
(3) Apply cover slip and examine under the microscope.

Ear wax sample

Ear wax can be collected and examined for the presence of *Otodectes cynotis*.

Method.

(1) Using a cotton bud, gently collect some of the ear wax from the external ear canal.
(2) Transfer the wax on to a microscope slide.
(3) Mix the wax with 2–3 drops of liquid paraffin or KOH and break up the clumps using a sterile needle so that the sample is not too thick.
(4) Apply a cover slip and examine under the microscope.

EXAMINATION FOR RINGWORM

As previously mentioned, dry plucked hairs can be examined microscopically. Obviously thickened hair shafts may be seen under low power and fungal spores can be seen with higher magnification (×100 or × 400). A drop of lactophenol cotton blue stain or 'Quink' ink may be added to the sample to make visualisation of the spores easier.

Ultraviolet (Wood's lamp) examination

A Wood's lamp uses an ultraviolet (UV) light to help detect certain strains of ringworm (50% of *Microsporum canis* show up this way). The dermatophytes fluoresce an apple-green colour when examined under the light.

Method.

(1) Switch on the Wood's lamp 5–10 minutes prior to examination to allow it to warm up.
(2) Examination should be carried out in a darkened room.
(3) Examination with a Wood's lamp can be done either by passing the lamp directly over the coat of the animal or by shining over plucked hairs for at least 3–5 minutes.

Remember that a negative Wood's light examination does not rule out ringworm because only a small percentage of ringworm are responsive to UV light and that other rare dermatophytes and certain bacterial infections can also fluoresce.

Fungal cultures

Hair pluckings can be collected and carefully inoculated, using sterile forceps, on to one of two media: Sabouraud's agar or dermatophyte test medium (DTM). DTM is available in commercial fungal culture kits for use in practices, with the name Dermafyte™. The unit containing the culture should be sealed with biohazard tape and kept at room temperature for 7–10 days. It should be checked daily for the appearance of a white colony and a red coloration of the medium, which indicates the presence of a dermatophyte. If there is no growth after 3 weeks, the sample can be considered negative for ringworm.

PRESERVING SKIN/HAIR SAMPLES

If it is not possible to examine the sample immediately, it may be stored until required. It may also be necessary to send the sample to an external laboratory for analysis in which case appropriate storage will also be required. It is important that samples that are not going to be examined immediately are not allowed to dry out. Potassium hydroxide or liquid paraffin may be used as the lubricant.

Table 38.1 Ectoparasites.

Insects Head, thorax and abdomen three pairs of legs	**Fleas** Macroscopic, brown laterally compressed body Move very quickly and can jump vast distances	*Ctenocephalides felis* Most common affecting dogs and cats *Ctenocephalides canis* *Spilopsyllus caniculi* (rabbit flea)
	Lice Macroscopic, whitish, brown, dorsoventrally flattened. Often attached to hairs. Lays eggs, 'nits', which are cemented to the hair shaft	*Felicola subrostratus* (Cat biting louse) Has two triangular points at head end resembling cat ears *Trichodectes canis* (dog biting louse) Blunt, flat head *Linognathus subrostratus* (dog sucking louse) Narrow head smaller than body
Arachnids (mites and ticks) Head and thorax combined (cephalothorax) and abdomen; four pairs of legs (some larval stages have three pairs of legs)	**Surface mites** Long legs, microscopic but slightly larger than subsurface mites	*Otodectes cynotis* (ear mite) round body, long hairs on hind legs, 'bells' on ends of front legs *Cheyletiella* spp. (fur mite) Roundish body with 'waist' C-shaped accessory mouth parts. Comb-like structures on ends of legs *Trombicula autumnalis* (harvest mite) Resemble orange small dots found around the ears and paws. Occur late summer, autumn. Only the larval stage is parasitic and has three pairs of legs
	Subsurface mites Short legs, microscopic	*Sarcoptes scabei* Round body short legs, burrows into the skin to lay eggs *Demodex* spp. Cigar-shaped body with short legs. Lives in hair follicle shafts
	Ticks Variety of different species. Can transmit disease	*Ixodes* spp. Body becomes engorged after feeding on host, usually greyish in colour resembling a wart

With the exception of smears, it is not advisable to send prepared microscope slides through the post as the sample is most likely to slip off the slide by the time it reaches its destination. Samples from skin scrapes can be placed into a 5 or 20 ml sterile universal sample pot along with the blade and sent to the laboratory. Always label the pot properly stating that a blade is in the pot to avoid any accidents. Samples of ear wax can be kept until ready or sent to the lab by cutting the cotton bud so that the piece with the sample on it

fits into a 5 or 20 ml universal sample pot. Special slide holders are available to send microscope slides in the post.

PARASITE IDENTIFICATION

An important role of the VN is assisting in identification of common ectoparasites affecting dogs and cats in practice. The main ectoparasites are either insects or arachnids (mites and ticks) (Table 38.1).

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SECTION 8

TRIAGE AND EMERGENCY PROCEDURES

INTRODUCTION

Under the Veterinary Surgeons Act 1966, VNs may render first aid in an emergency for the purpose of saving a life or relieving pain or suffering in an animal. According to the guidelines from the Royal College of Veterinary Surgeons this is restricted to what is necessary to save an animal's life or stop pain or suffering until the veterinary surgeon can be contacted. It is therefore essential that qualified and listed nurses have the expertise and competence to carry out first aid procedures appropriate to the needs of the patients and that they are fully aware of all procedures that may be rendered in an emergency situation to fulfil the aims of first aid. This section covers the triage and immediate first aid requirements for a number of different conditions. Emergency procedures are covered, including placing intravenous lines.

Chapter 39

Initial Assessment

These notes list the action VNs should take in the event of being responsible for an emergency case arriving at the clinic.

The aims of first aid are:

- preservation of life;
- prevention of suffering;
- prevention of the deterioration of the patient's condition;
- relief of acute conditions.

Rules for the VN:

- do not panic;
- maintain airway;
- control haemorrhage;
- contact the veterinary surgeon ASAP!

TELEPHONE CALLS

What to ask

- Nature of injury and extent of injury.
- Time of incident.
- Breed, age, sex of animal.
- Pre-existing conditions and current veterinary treatment: diabetic? cardiac? epileptic?
- Caller/owner's name, address and contact telephone number.
- Their estimated time of arrival.

Above all, stay calm and reassure the caller.

Advice to callers for handling and transportation of the injured animal

- Avoid further dangers to the animal or the caller (warn oncoming traffic, approach the animal carefully) – shocked and injured animals can become aggressive.

- Is the animal breathing? Clear airway, pulling tongue forward and remove debris from the mouth if possible without putting a hand in the animal's mouth, gently extend the neck unless there is obvious trauma.
- Control haemorrhage – apply constant pressure to area using material that is as clean as possible.
- Keep the animal warm using coats, blankets, etc.
- Advise caller about moving the animal – use a coat, blanket or board to make a stretcher following a road traffic accident (RTA) or multiple trauma.
- Use an aerated box or cat basket for cats and small dogs.
- Ensure the animal is secure and unable to escape.
- Keep the animal as immobile as possible ensuring the traumatised area is uppermost.
- Calm and reassure the animal.

PREPARATION AT THE VETERINARY CLINIC

- Inform all other veterinary staff of the nature of the accident.
- Prepare kennelling – blankets and heat pads.
- Prepare equipment for intravenous fluid therapy – warm fluids.
- Prepare oxygen administration equipment.
- Prepare emergency drugs.
- Ensure X-ray developing machine is switched on.

EXAMINATION OF THE PATIENT ON ARRIVAL AT THE CLINIC

Carry out the ABC procedure.

- A: Check and clear **airway**: pull tongue forward, remove debris using swabs or suction, intubate if possible or perform tracheotomy. For obstructions in the larynx, perform Heimlich manoeuvre.
- B: Check **breathing**: institute artificial respiration if necessary. If the animal is breathing, observe respiratory pattern – is inspiratory distress present? Is there evidence of obstruction? Cover open wounds to throat or chest.
- C: Check **circulation**: is there a heartbeat? Check mucous membrane colour. If no heartbeat then start cardiopulmonary cerebral resuscitation (CPCR) (see Chapter 40).

If immediate resuscitation is not necessary, a primary survey may be performed. Four body systems are evaluated: respiratory, cardiovascular, neurological and renal (Table 39.1). These systems are critical to the animal's survival; repair of wounds and fractures can be completed once the animal is stable.

Table 39.1 Assessment of body systems.

Respiratory	Observe for signs of respiratory compromise: dyspnoea, tachypnoea, pale or cyanotic mucous membranes. Is the patient standing or lying in a particular position to enhance respiration. If the airway is patent, then oxygen may be administered
Cardiovascular	Is femoral or peripheral pulse palpable? Measure capillary refill time. Listen for heart arrhythmias or abnormal sounds. Circulatory collapse leads to shock which if not treated will almost certainly result in death. Control haemorrhage; cover open wounds using pressure with sterile dressing. Take core body and extremity temperature and take steps to maintain normothermia
Neurological	Observe animal's mental state. Can it stand and walk? Check cranial nerve function. Neurological signs may also be result of metabolic dysfunction
Renal	Evaluation of kidney and urinary function is necessary but less easy to assess. Palpation of the bladder and, later, monitoring of urinary output are part of monitoring this particular system. Fluid administration to replace lost fluids and maintain circulation is required in the majority of cases

Table 39.2 Monitoring the critical patient.

Demeanour	Whether the patient is bright and alert or lethargic, depressed and unresponsive. It is important to recognise whether a patient is normally quiet or whether it is a result of its condition
Temperature	An increase in the animal's body temperature may indicate infection whilst a decrease may indicate shock. Check the extremities (paws and ears) to see if they feel cold. This may indicate poor peripheral perfusion often seen in cardiac disease
Pulse	Measurement of the pulse gives an indication of the heart rate. Remember to check peripheral pulses as well as central pulse. This will give an idea of how good the circulation is
Respiration	Record the rate as well as the depth and rhythm. Does the animal require assisted ventilation or oxygen?
Mucous membrane colour	The colour of the mucous membranes can give an indication of various conditions. Cyanotic colour and tachypnoea indicate hypoxia. A pale colour indicates shock, anaemia or haemorrhage. Yellow mucous membranes indicate hepatic dysfunction
Capillary refill time	A prolonged capillary refill time indicates poor peripheral perfusion. Normal time should be less than 2 seconds
Urine output	In critical care patients, urine monitoring is important to assess kidney function. Closed urinary catheterisation ensures accurate recording. Assessment of urination without catheterisation or cystocentesis is less accurate but a record should be made of the number of times the animal has urinated, whether in its cage, litter tray or when taken outside
Pain	Animals in shock may not show specific signs of pain. Different breeds react differently. It is essential to control pain not only from a welfare view but also the influence pain has on physiological parameters. Pain also has physiological effects (vasoconstriction to vital organs, reduced wound healing and inappetance)
Appetite	Critical care patients are often inappetent and require assisted feeding techniques. Animals that are inappetent for long periods of time become cachexic, not only losing weight but also muscle mass as the body tries to assimilate energy from its own sources

Table 39.3 Monitoring equipment.

Electrocardiogram (ECG)	Continued monitoring of the electrical activity of the heart is necessary to ensure normal functioning and to indicate deterioration. Beware of a condition called pulseless electrical activity (PEA) where a normal trace may be seen without actual contractions. Peripheral pulses must also be palpated to check heart function
Pulse oximeter	Pulse oximeters are used to determine the oxygen saturation of haemoglobin. They can be useful to detect pulses also. Care must be taken with interpretation as 100% reading does not always mean there is adequate ventilation
Capnograph	In cardiac arrest pulmonary circulation decreases and carbon dioxide is not delivered to the alveoli for removal from the body. Exhaled carbon dioxide is a good measure of cardiac and respiratory resuscitation
Blood gases	Arterial blood gas analysis gives a definitive assessment of lung function
Blood tests	Packed cell volume, glucose and electrolyte measurement are reasonably quick tests that can be performed in house

A secondary survey may then be carried out ensuring a full clinical examination. The mnemonic below is a useful reminder.

A Airway – examine oral cavity, pharynx and neck.
C Cardiovascular system – monitor and record heart rate, pulse rate and quality.
R Respiratory system – observe respiratory movements, monitor and record respiratory rate and depth
A Abdomen – examine and note any bruises, punctures and protrusions.
S Spine – neurological examination.
H Head – examine all parts, eyes, ears, nose, teeth and mouth.
P Pelvis.
L Limbs – examine for evidence of fractures, wounds.
A Arteries – external arterial haemorrhage will have been picked up earlier. Is there internal bleeding?
N Nerves – check reflexes to assess level of consciousness: pedal withdrawal reflex, pupillary and palpebral reflexes.

MONITORING THE CRITICAL CARE PATIENT

Depending on the severity of the patient's condition, monitoring may be performed from anything from three times an hour to three times a day. The details should be recorded accurately so that trends may be noted. See Table 39.2.

Monitoring equipment

Baseline parameters should be recorded and there are a number of useful pieces of monitoring equipment which are essential to the critical patient. See Table 39.3.

Chapter 40

Emergency Procedures

It is important that the VN is able to carry out various essential emergency procedures.

CARDIOPULMONARY CEREBRAL RESUSCITATION

Cardiopulmonary arrest is the cessation of ventilation and effective circulation. Irreversible brain damage starts to occur after 3 minutes without an oxygen supply and it is described as a '3-minute emergency'. The VN must be fully competent in the actions that must be taken in order for cardiopulmonary cerebral resuscitation (CPCR) to be effective. There is no time to consult manuals and it is obviously not a task that can be practised easily. The main factors that control the degree of success include:

- preventing the factors that predispose animals to cardiac arrest;
- recognising the signs of cardiac arrest;
- performing the appropriate CPCR techniques in the correct sequence.

Factors predisposing animals to cardiac arrest

If VNs are aware of the various causes of cardiac arrest they will not only be more likely to be alert to the problems when nursing these high-risk patients but also more likely to take appropriate steps to prevent the situation developing into arrest. Predisposing factors include:

- drugs (e.g. anaesthetic drug overdose, digoxin, etc.);
- toxins (e.g. endotoxin release during sepsis depresses cardiac function and causes arrythmias);
- hypoxia;
- hypovolaemia/hypotension;
- acid–base imbalances (extremes in blood pH cause myocardial irritability and contractility);
- electrolyte imbalances (abnormal potassium levels produce changes in heart function);
- extremes of temperature (changes seen with hypothermia dramatically increase risk of cardiac arrest).

Signs of cardiac arrest

- No pulse.
- No heart beat.
- Grey/cyanotic mucous membranes.
- Dilated pupils, eye centrally fixed.
- Increased capillary refill time (CRT).
- No respiratory movements or agonal gasps.
- No bleeding at surgical site.
- ECG – asystole, ventricular fibrillation or pulseless electrical activity.

Procedure

It is necessary to clarify whether to commence CPCR in the first place as it is a costly procedure and takes time and expertise. Several trained personnel are required throughout the resuscitation procedure to attend directly to the animal and also to obtain drugs and equipment. High-risk cases should therefore be discussed at the time of admission to the hospital or before surgery and those where CPCR is not to be carried out should be identified. (NFR – Not For Resuscitation or DNR – Do Not Resuscitate signs can be placed on kennels of those cases when appropriate.) For those cases where CPCR is indicated, it is essential that the arrest is identified as soon as it has happened and then appropriately acted upon straight away. Remember that this is a '3-minute emergency'. The procedure is described in Table 40.1.

Signs of effective CPCR include:

- palpation of a femoral pulse during cardiac compression;
- constriction of the pupil;
- improvement in the colour of the mucous membranes;
- ventromedial rotation of the eye;
- normal ECG pattern returning.

Signs of continued improvement include:

- return of spontaneous respiration;
- return of palpebral eye reflex;
- vocalisation;
- lacrimation;
- return of movement, righting reflexes.

RESUSCITATION OF THE NEONATE

Receiving the neonate from the surgeon

The surgeon will remove the puppy from the uterine horns and want to pass it on quickly so that he or she can continue with the surgery. The VN holds a towel in both hands and allows the surgeon to 'drop' the neonate into the

Table 40.1 Procedure for CPCR.

Call for help and note the time	
A	**Airway** Clear airway – remove mucus and debris, pull tongue forward Intubate if possible
B	**Breathing** **Intubate and ventilate with 100% oxygen** This maybe via a Bain or Ayre's T-piece breathing system or an AMBU bag connected to the oxygen supply. Ventilate between 6 and 12 times a minute taking care not to overinflate. The bag should be compressed quickly allowing short inspiration and longer expiration If intubation is not possible, then mouth to nose resuscitation is necessary (wear face mask or cover nose with swab)
C	**Circulation** **Perform chest compressions around 80–100 per minute** Place animal in right lateral recumbency, with a sandbag under the thorax Compress over 6th intercostal space using both hands and keeping arms locked Use force appropriate to the size of the animal In larger dogs (>20 kg) increasing the intrathoracic pressure increases the pressure within the intrathoracic vessels and can be more effective than the cardiac pump. It involves compressing the cavity Concurrent ventilation can assist compression Hold compression for a brief time to maximise elimination of blood from the heart Check pulse regularly – ideally another person should check the pulse for the effectiveness of the compression Open cardiac massage may be necessary
D	**Drugs** Administer drugs under veterinary surgeon's supervision. Possible drugs are: **Adrenaline** – stimulates sinus rhythm, increases cardiac contractility and constricts peripheral vessels. Give intravenously or intracardiac **Lignocaine** – if heart is fibrillating slows heart rate down **Sodium bicarbonate** – to treat acidosis The use of each drug during CPCR is dependent on the abnormality and ECG readings. The drugs can be administered by the following routes, in order of preference: **Cranial vena cava** (via a jugular catheter) **Intratracheal** (squirted into the trachea via a long catheter, through and beyond the endotracheal tube). It should be noted that higher doses than normal are required and the drugs should be added to 0.6 ml/kg of sterile saline **Intraosseous** especially in young animals **Peripheral veins** (slow reaction to drugs given by this route) **Intracardiac** (potential hazards including intramyocardial injection inducing fibrillation or tearing of the lung leading to pneumothorax) Have a chart available with calculated drug doses for animals in increments of 2–5 kg
E	**Electrocardiogram** Set up ECG. Note abnormal rhythms and report to veterinary surgeon

Table 40.1 *Continued*

Call for help and note the time	
F	**Fluids** Set up intravenous fluids (crystalloid or colloid). The aim of fluid administration is to increase the circulating blood volume quickly. For this reason, colloids are often the fluid of choice and should be injected as rapidly as possible by syringe at a volume of 25 ml/kg (cats) and 50 ml/kg (dogs). If colloids are not available, compound sodium lactate (Hartmann's) can be given at the same rate by squeezing the bag. After the initial infusion, fluids can be administered at a shock rate of 20 ml/kg/hour

open towel without contaminating the surgeon. The VN must then take the following steps quickly and efficiently.

(1) Using the roughness of the towel, break open the fetal membranes from around the neonate's head and body.
(2) Wipe the mouth and nose area with the towel to remove most of the fluid.
(3) Check to ensure a heart rate. If present continue steps 4–12. If absent, follow resuscitation procedure.
(4) Use a cotton bud to gently prise open the mouth and use it to wipe away the fluid inside the mouth.
(5) Use the towel to rub the neonate to stimulate respiration.
(6) Respiration should now occur. Usually, a sharp, short inhalation is followed by short period of apnoea – keep rubbing neonate to stimulate. Remember, it will still have the cardiorespiratory depressant effects of the anaesthetic agents.
(7) By now, the surgeon is probably ready to pass on another neonate so follow the above procedure.
(8) Place the first neonate into a box or large litter tray with hot water bottle/heated corn pad and clean bedding and keep checking it whilst sorting out the next neonate.
(9) Place new neonates next to the one before and, at every opportunity, keep rubbing them to help stimulate them, help keep them warm and dry off their fur.
(10) Keep following the procedure shown above until all of the litter has been delivered.
(11) If there are more people around, e.g. the owner, get them to rub the neonates with a towel.
(12) When all the neonates are delivered safely, tie off the umbilicus with gut suture material about 1 cm from the abdominal wall and cut just distal to the ligature.

Action in the event of apnoea or respiratory depression

If there is a heartbeat but the neonate does not start breathing by stage 6 above, the next few steps should be taken.

(1) Hold neonate so that head is lower than the rest of the body to enable fluid to drain out. Reswab mouth using cotton bud or pipette
(2) Administer respiratory stimulant, e.g. doxopram hydrochloride (Dopram-V™). This is most frequently given orally by placing 1–2 drops under the tongue. Do not overdose.
(3) Gently blow down the nose and mouth (avoid direct contact) to provide oxygen.
(4) Continue to stimulate the neonate by rubbing its entire body with the towel.
(5) Continue to check for the presence of a heart beat.
(6) Continue until regular respiratory efforts are maintained.

Action to take if there is no heart beat

In some cases, the neonates have already died before the Caesarean surgery, and cardiovascular resuscitation is not indicated. However, occasionally, there may not be a heart beat due to the depressant effects of the anaesthetic agents and cardiac massage can be performed.

(1) Place thumb and first finger on either side of the thorax over the position of the heart (if unsure, use point of elbow as a guide).
(2) Squeeze the thorax using firm, fast and regular movements (the neonate's normal heart rate is over 200/min, so there is no way this rate can be achieved – aim to squeeze as quickly as possible in an effective way).
(3) Every 15 seconds or so, check for the presence of a heart beat and stimulate respiration as detailed above.
(4) Check the neonate for obvious abnormalities that may be hindering attempts.
(5) Continue until success or otherwise instructed to stop.

INTRAVENOUS CATHETERISATION

Gaining venous access is one of the most important procedures in emergency care. Drugs and fluids can then be administered as necessary. Table 40.2 lists catheter sizes.

Equipment required:

- clippers with number 40 or 50 blade;
- cleansing swabs (chlorhexidine scrub or other appropriate surgical scrub solution);
- swab soaked with isopropyl alcohol;
- additional gauze swabs;
- 2.5 cm adhesive tape cut to lengths that will encircle catheterisation area;
- padding bandage material;
- conforming bandage;
- scissors;
- syringe containing 2 ml heparinised saline;
- appropriate size/type catheter (see Table 40.2);
- T-connector, three-way tap or bung.

Table 40.2 Recommended catheter sizes.

Animal	Cephalic catheter	Jugular catheter
Cat or dog <10 kg	22–24 gauge	20–22 gauge
Dog 10–20 kg	20–22 gauge	18–20 gauge
Dog >20 kg	18–20 gauge	14–18 gauge

Cephalic catheterisation

Collect equipment and wash hands before placing catheter (see method in Fig. 40.1).

Method.

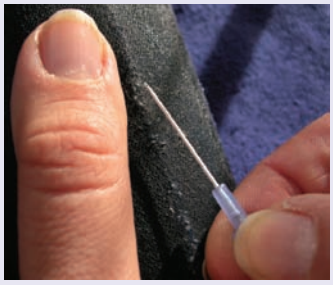
(1) Collect the equipment and flush the T-connector, three-way tap or bung with heparinised saline.
(2) Ensure the patient is adequately restrained.
(3) Clip a small square over the cephalic vein.
(4) Aseptically prepare the site with antiseptic solution and spirit.
(5) Wash hands.
(6) Ask the assistant to raise the vein.
(7) Identify the vein without touching the site. Stabilise the vein and insert the catheter with the bevel facing upwards.
(8) As the catheter enters the vein, blood will appear in the hub of the catheter.
(9) Hold the stylet and advance the catheter into the vein up to the hub.
(10) Ask the assistant to stop raising the vein and to place their thumb 2–3 cm above the catheter entry site to prevent blood flow.
(11) Remove the stylet and attach the connector (T-connector or three-way tap).
(12) Secure the catheter by placing a strip of tape under the hub of the catheter then around the leg and back over the top of the hub. Further strips of tape may be placed around the connector.
(13) The catheter should then be covered with a padding layer and outer cohesive bandage.

Jugular catheterisation technique (using a 'through-the-needle' jugular catheter)

Method.

(1) Collect all necessary equipment and flush the T-connector with heparinised saline.
(2) Assistant restrains the animal in lateral recumbency, extending head to enable access to the jugular.
(3) A small sandbag placed under the neck will enhance visualisation and catheterisation of the jugular.

Fig. 40.1 Placing an intravenous catheter.

METHOD
Collect the equipment and flush the T-connector, three-way tap or bung with heparinised saline
Ensure the patient is adequately restrained
Clip a small square over the cephalic vein
Aseptically prepare the site with antiseptic solution and spirit
Wash hands
Ask assistant to restrain patient and raise the vein
Remove catheter from packet (do not touch any part of the needle)
Identify the vein without touching the site. Position thumb so that it is just lateral to the vein to help stabilise the vein (Fig. 40.1a)  40.1a
 40.1b Holding catheter in right hand, insert the needle through the skin at a 20–30° angle, bevel facing upwards (Fig. 40.1b)
Advance catheter one third of way into vein – blood should appear in hub when it enters the vein (Fig. 40.1c)  40.1c

Continued

40.1d

Stabilise the stylet hub with left hand (Fig. 40.1d)

With the other hand, gently slide and advance catheter into vein up to the hub

Observe to ensure blood is still flowing freely from catheter hub

Continue holding catheter in place to prevent dislodgement

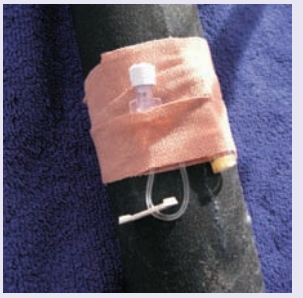
Ask assistant to stop raising vein and move thumb over skin over catheter to help reduce blood flow

Remove catheter stylet and attach pre-filled T-connector

Secure catheter with tape appropriately:

- First strip placed *underneath* the catheter and then wrapped over the skin insertion site and catheter hub
- Second strip placed under the T-connector and wrapped around the limb and over the T-connector

Loop made in T-connector tubing and third strip placed to secure loop in place. A sterile transparent adhesive dressing may be placed over the catheter site to prevent infection (Fig. 40.1e)



40.1e

Flush catheter with small quantity of heparinsed saline

Apply final bandage layer over catheter and connector

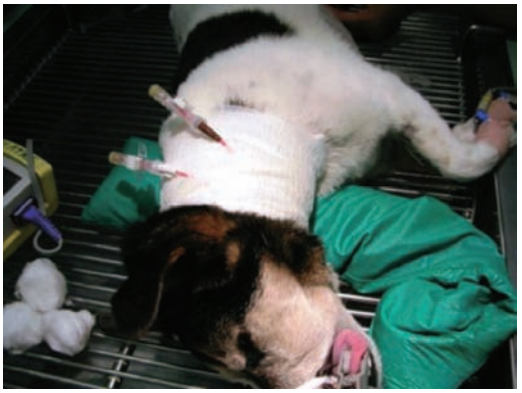


Fig. 40.2 Bandaging the jugular catheter.

(4) Clip hair over the jugular area.
(5) Wash hands.
(6) Aseptically prepare the site using surgical solution.
(7) Rinse soap suds off with swab soaked in isopropyl alcohol.
(8) Wash hands and put on sterile surgical gloves.
(9) Raise the vein by applying pressure with the thumb to the jugular groove.
(10) Hold the needle firmly in one hand, and insert through the skin in a caudal direction towards the thoracic inlet.
(11) Insert the needle through the skin before attempting to enter into the vein.
(12) Stabilise the jugular vein with the free hand and with one sharp movement, pass the catheter into the vein.
(13) When blood appears in the hub and lumen of the catheter advance the catheter 1 cm further.
(14) Still holding the catheter in the right hand, thread the catheter into the vein by pushing it through the metal needle within the plastic sleeve.
(15) Disconnect the plastic sleeve from the needle.
(16) Take the metal needle out of the skin and place the protective plastic needle guard over the needle and snap closed.
(17) Place injection cap on to catheter.
(18) Flush catheter with heparinised saline.
(19) Bandage the catheter by initially placing a sterile dressing over the insertion site and then covering with padding and cohesive bandage, ensuring it is not too tight (Fig. 40.2).

Over-the-wire technique

The Seldinger technique uses an over-the-wire catheter and a guide wire. An introducer catheter is placed into the jugular vein and a guide wire is passed through this into the vein. The catheter is removed and a dilator is threaded over the wire to enlarge the diameter of the insertion site. This is then removed

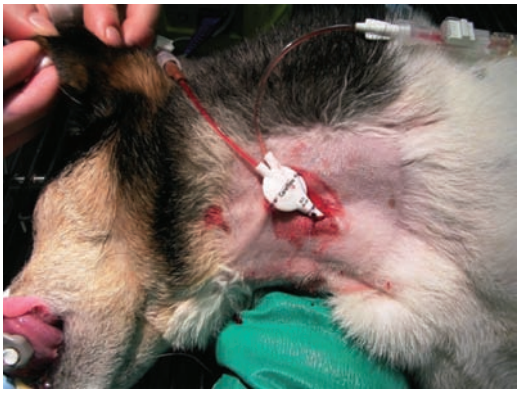


Fig. 40.3 Placement of a jugular catheter.

and the jugular catheter threaded over the guide wire and into the vein. The catheter is then secured by suturing the wings to the skin (Fig. 40.3).

Useful tips

In thick-skinned animals, or animals in shock or dehydration, it may be advantageous to make a small 'nick' through the skin just above the vein to help visualise and catheterise the vein more easily. The skin is pulled away from the vein and a small cut is made in the skin using a 20 gauge needle or number 11 surgical blade. The catheter can then be inserted through the skin at this point.

Effective restraint of the patient is essential for successful catheter placement – take time to make the animal more at ease and provide lots of vocal reassurance. If possible, try to place catheters before administration of sedatives such as acepromazine or medetomidine, which dramatically drop blood pressure thus making catheterisation more difficult.

Appropriate dilution of heparinised saline may be made by adding 0.1 ml of heparin (1:1000) to 100 ml of sterile saline.

THE HEIMLICH MANOEUVRE

The Heimlich manoeuvre is used for removal of an obstructive foreign body in the larynx. The technique employed depends very much on the size of the animal.

Method.

(1) Suspend patient upsidedown by the hindlegs. Large dogs may be hung over a table.
(2) Place a hand on the animal's back and one hand just below the rib cage.
(3) Compress inwards and upwards to increase the intrathoracic pressure. For large dogs use both hands under the ribcage.
(4) The patient should cough and expel the foreign body.

If the obstruction is not dislodged – an emergency tracheotomy may be required. Try feeding a urinary catheter alongside the obstruction and attach it to the oxygen supply. A urinary catheter may be adapted and kept specifically for this use by attaching an endotracheal adaptor to the end and securing it in place. This will ensure it fits the oxygen supply.

TRACHEOTOMY

A tracheotomy is performed to relieve upper respiratory obstructions in order to alleviate asphyxiation. In an emergency situation any sharp instrument can be used to make the tracheotomy hole. A large-bore needle (14–16 gauge) can be inserted between two cartilage rings to provide ventilatory access.

Method.

(1) Dampen the hair over the trachea with surgical spirit.
(2) Make an incision caudal to the larynx (between 3rd and 4th tracheal ring).
(3) Place stay sutures in the 3rd and 4th ring to keep the insertion site open.
(4) Insert a endotracheal tube (50% diameter of trachea) directing downwards towards the lungs.
(5) Attach to oxygen supply via breathing system or AMBU bag.
(6) Close the subcutaneous tissue and skin around the tube.
(7) Secure with umbilical tape around the neck.

Usually this is a temporary measure until the obstruction is removed.

Manual positive pressure ventilation

Indications include:

- resuscitation in the event of respiratory/cardiac arrest;
- during intrathoracic surgery;
- following administration of neuromuscular blocking agents;
- respiratory inadequacy.

Equipment needed:

- intubated animal;
- 'Ambu' resuscitator self-inflating bag or a suitable anaesthetic breathing system, e.g. Ayres T-piece or Bain;
- oxygen supply (anaesthetic machine).

The rate should be around 6–10 breaths per minute. The volume of oxygen should be sufficient to produce good chest movements (excessive pressure must be avoided to prevent lung damage) at around 8–12 ml/kg. The inspiratory phase should last only about one third of the time of the total ventilatory cycle, i.e. there is a short inflation followed by a longer expiratory pause. This facilitates venous return. A time ratio of 1:2 is recommended.

Table 40.3 Methods of oxygen supplementation.

Method	Use
Oxygen mask	Good for emergency, short-term use High flow rates (10 l/min) to prevent CO_2 build-up Patient intolerance of mask
Oxygen cage	Can be difficult to observe and manage patients in oxygen cages Cage can be warmed/humidified Inefficient (oxygen lost each time door opened) High flow rates required (initially 10 l/min, reducing to 5 l/min for maintenance) Expensive
High flow-by	Tube fed into cage and placed by patients face Useful if animal is open-mouth breathing High flow rates
Intratracheal catheter	A 14 gauge intravenous catheter is placed aseptically in the trachea and then attached to an oxygen supply at a rate of 100 ml/kg/min. Oxygen should be humidified Low flow rates Very efficient May be difficult to restrain animal without stress Useful if larynx/pharynx is obstructed Needs time to set up – no good for emergency use
Nasal catheter (or nasal prongs)	A 5 Fr or 8 Fr soft feeding tube is inserted into the nasopharynx via the nostril. The tube is secured using butterfly strips. The oxygen should be humidified and administered at a rate of 100 ml/kg/min Good for long-term use Low flow rates
Elizabethan collar	Place 'cling film' over the front of an Elizabethan collar leaving a small gap at the bottom. The oxygen tube is placed through the collar at the back of the patient's neck. Flow rates of 2–4 l/min should be used. Cheap and easy to set up Well tolerated especially by cats Reasonably efficient

Technique.

(1) Allow the bag on the circuit to inflate by temporarily partially closing the adjustable pressure limiting (APL) valve.
(2) Inflate the lungs by squeezing the bag. Observe chest wall movement to ensure overinflation does not occur.
(3) Open the valve and repeat cycle.

In cats it is advisable to keep the endotracheal tube uncuffed to avoid overinflating the lungs.

OXYGEN THERAPY

Oxygen therapy is required when there is inadequate tissue oxygenation (hypoxia). This can occur due to dyspnoea or circulatory failure (shock). It is important to remember that hypoxic animals do not necessarily show clinical signs until things are extremely severe and, for this reason, oxygen therapy should be considered for any patient with reduced lung volume or poor peripheral perfusion. Do not wait until an animal exhibits signs associated with hypoxia (e.g. dyspnoea, cyanosis, cold extremities, cardiac rate changes, etc.) before starting therapy.

The aim of oxygen therapy is to increase the amount of oxygen in the blood by increasing the arterial oxygen tension and this can be achieved using the methods listed in Table 40.3. The condition and demeanour of the patient need to be considered and the method of administering oxygen should take into account the most appropriate method for the patient.

Notes on humidification of oxygen

In the normal patient, air passing through the airways is warmed, moistened and filtered by the epithelial cells of the nasopharynx. Humidification is essential when oxygen therapy is being delivered using a system which bypasses the patient's own humidification system. Commercial bubble humidifiers are available or alternatively the oxygen can be administered by passing the flow through a bottle of warmed saline. The tube from the oxygen source bubbles through the saline. The tube going to the patient lies above the level of the saline.

STOMACH TUBING FOR GASTRIC DILATATION

It is important to try to relieve the excess gas that builds up as quickly as possible.

Method.

(1) Measure the tube from the caudal end of the ribs to the nose.
(2) Place a mouth gag or a roll of bandage in the animal's mouth so that the tube can be fed through the middle.
(3) Lubricate the end of the tube and gently insert it into the oesophagus allowing the animal to swallow.
(4) As soon as the tube enters the stomach, gas and stomach contents may come back up the tube.
(5) If the stomach is twisted it may not be possible to pass the tube in which case surgery is necessary.
(6) After the stomach is decompressed it may be lavaged several times with water or a water and charcoal solution.
(7) Observe the animal closely for further signs of gastric distension.
(8) A measuring tape or piece of non-conforming bandage may be used to measure the abdominal circumference at frequent intervals to check for enlargement.

Chapter 41

Blood Transfusions

Blood transfusions are integral to the support care of critically ill and anaemic patients and can save the lives of many patients. The VN must be competent in all aspects of practical collection and administration and confident at monitoring the patient receiving the blood.

The aims of blood transfusion are:

- to restore oxygen-carrying capacity of blood;
- to replace various blood components (plasma proteins, platelets, clotting factors).

Anaemia is the main indication for a blood transfusion in veterinary practice. This may be a result of acute blood loss from trauma or a chronic, often immune-based condition. It is recommended that a transfusion should be administered if the patient shows clinical signs of anaemia and the packed cell volume (PCV) is less than 10% or if there has been a rapid decrease in the PCV to less than 15–20%.

BLOOD COLLECTION

Choosing a donor

Canine blood donors should be healthy (no current medication except antiparasiticides), vaccinated and wormed against common endoparasites. They should weigh at least 25 kg to allow collection of one unit using human collection bags. As far as age limits, this can depend on the breed but donors are usually 1–8 years old.

Feline blood donors should also be healthy, vaccinated and wormed against common endoparasites. The cats are usually aged 1–8 years and weigh at least 4 kg. Most feline donors require sedation for collection.

Note.

- Greyhounds have a higher PCV (48–66%) than most breeds therefore are very useful as donors.
- Giant breeds may be able to give more than 1 unit.
- A minimum of 4–6 weeks should be left in between each donation.
- Routine PCV measurements should be taken prior to any donation.

Blood collection containers

Dogs

Human collection bags provide 1 unit (450 ml blood + 50 ml anticoagulant) and contain anticoagulants to prevent clotting and to preserve the blood. Anticoagulants used in human blood bags are:

- CPDA (citrate–phosphate–dextrose–adenine);
- ACD (acid–citrate–dextrose).

Cats

Blood for feline transfusion can be collected into a 50 ml syringe containing 7 ml of ACD or CPDA (withdrawn from a human blood collection bag). The ratio is usually 1 ml of ACD to 7–9 ml of blood and 1 ml of CPDA to 7 ml of blood.

Amount to collect

No more than 20% of blood volume should be collected. As a general rule, up to 10% of a donor's blood can be taken without adverse effects on the donor. Up to 20% of blood volume can be obtained but it is recommended that intravenous fluids should be administered during and after collection at a rate of twice maintenance requirements.

The total blood volume is 90 ml/kg for dogs and 60 ml/kg for cats. Therefore the recommended volume limits are:

- dogs 18 ml/kg;
- cats 10–12 ml/kg.

Method of collection

Find a quiet room and at least three people:

- assistant to restrain animal;
- assistant to hold blood bag and mix the collected blood;
- person to carry out venepuncture.

The owners may wish to stay with their animal and they may be useful to help restrain and calm the pet during the procedure.

(1) Sedate donor animal if necessary (avoid circulatory depressants such as medetomidine).
(2) Collect all necessary equipment: clippers, collection bag, alcohol swabs and surgical scrub, etc.
(3) Assistant restrains donor for jugular venepuncture: giant breeds sitting on floor; smaller dogs sitting or in lateral recumbency on a table; cats in lateral recumbency on table.

(4) Clip and aseptically prepare site over jugular.
(5) Raise the jugular vein and aseptically place the blood bag needle into the jugular vein (or 21 gauge butterfly catheter prefilled with anticoagulant and attached to 50 ml syringe in cats).
(6) Hold the collection bag lower than animal (gravity enhances flow rates) or apply very gentle suction to the syringe (excess suction will collapse the vein).
(7) Second assistant gently agitates the bag or the operator inverts the syringe regularly throughout collection to thoroughly mix the blood with the anticoagulant.
(8) Check the bag periodically to ensure that the blood is still flowing.
(9) Continue until the desired quantity of blood is obtained. The bag can be weighed to find out approximate measurement because 1 ml of blood weighs approximately 1.053 g. A full bag should weigh approximately 500 g.
(10) Remove the jugular needle and the assistant should apply pressure for at least 5 minutes, over the venepuncture site to prevent a haematoma forming. A dressing may be applied around the neck to keep the swabs in place for a couple of hours.
(11) Allow the tubing to refill with blood and tie or clamp the tubing in several places along its length – this allows for sampling the blood for cross-matching.
(12) Label bag with the donor's name, date of collection, and date of expiry.

Advise the owners to keep the donor animal calm for at least 30 minutes and rested for the remainder of the day. They should avoid putting a lead and collar on for as long as possible and can feed the animal after the procedure. Cats should be given 30 ml/kg intravenous fluids over the next 3 hours and offered food and water once they have recovered from the sedation.

STORAGE OF WHOLE BLOOD AND COMPONENTS

Fresh whole blood must be used within 8 hours of collection. Blood taken from a donor and not used immediately needs to be stored in a refrigerator at 4°C. The blood bags should be placed inside another plastic bag to avoid contamination of the fridge should spillage occur. The bag should be kept upright to maximise gas exchange, helping to preserve the viability of the red cells. Ensure all blood collected is labelled with the donor's details, date of collection and date of expiry. If the blood transfusion to the recipient is required for platelet replacement it should be done within 6 hours of collection. For red blood cell (RBC) replacement, the collected blood should used within 4 weeks.

Plasma products should be frozen at -20°C. Place a rubber band around the bag creating a 'waist' and place in the freezer. Once the plasma has frozen the band is removed. If the plasma thaws and refreezes the 'waist' disappears and the plasma should not be used.

Packed red cells are separated from the plasma by centrifugation. Using collection bags which have satellite bags aids this separation process. The

cells may be stored at 4°C for approximately 20 days, longer if preservatives are added.

ADMINISTRATION OF THE BLOOD OR COMPONENTS

Donor/recipient blood matching

Before transfusion is carried out, a cross-matching test or blood typing analysis should be carried out to ensure that the donor blood is compatible with the recipient's blood, thereby reducing the likelihood of a transfusion reaction. Blood matching is especially vital in feline transfusion or second transfusions in dogs, because fatal reactions can occur if incompatible blood groups are mixed. There are two methods of ensuring that donor and recipient blood is compatible:

- blood typing;
- cross-matching.

Blood typing

Dogs

There are eight different blood groups that have been identified in dogs: dog erythrocyte antigen (DEA). Ideal blood groups are DEA 1.1 (negative) and DEA 1.2 (negative), which account for approximately 40% of the dog population. These blood groups should ideally be used whenever possible in transfusions, because they do not produce antibodies against blood and can be used safely in dogs that have previously received blood transfusions.

Cats

There are three blood groups in cats:

- A group – most common, generally domestic shorthair and Siamese types;
- B group – mainly domestic longhair and exotics;
- AB group – rare.

Although blood type A is the most common, the frequency of types A and B varies geographically and among breeds. Like humans, cats have significant naturally occurring antibodies against the blood type antigen that they lack. It is essential that blood typing or cross-matching be carried out first to prevent potentially fatal reactions.

Cross-matching

Cross-matching is a test that is performed to assess whether the donor and recipient serum antibodies will mix and not destroy each other. Blood samples must be obtained from both the donor and the recipient and collected into heparin and EDTA sample tubes. Samples of plasma and washed blood cells are mixed on slides and the samples examined for evidence of agglutination and possible haemolysis of the blood cells.

Transfusion quantities

The aim of transfusion is to increase the recipient's PCV to above 20–30%. This should reverse the signs of anaemia and alleviate the life-threatening condition of the patient.

The following formula can be used to estimate the volume of blood needed for transfusion:

$$\text{Volume (ml)} = 85\ \text{(dog) or } 60\ \text{(cat)} \times \text{bodyweight (kg)} \times \frac{\text{(desired PCV} - \text{actual PCV)}}{\text{donor PCV}}$$

Example:

A 15 kg dog, has a PCV of 10% and the aim of transfusion is to get up to a PCV of 25%. The PCV of the donor dog is 40%.

$$85 \times 15 \times (25 - 10)/40 = 478 \text{ ml}$$

Therefore, 500 ml of whole blood should be given to this patient to achieve the desired PCV.

Method of administration

Blood products may be administered via any appropriate intravenous route or, in the case of very young animals, the intraosseous route may be used. Freshly collected blood will be at the correct temperature for immediate administration. Refrigerated blood and frozen plasma will need to be warmed to at least room temperature before administration. This should be achieved slowly to avoid clotting and red cell lysis. Blood warmers (water baths and dry heat blood warmers) are available.

Packed red cells are administered after being resuspended in or co-administered with physiological saline. Normal fluid administration sets are not suitable. Special blood administration sets are available for attachment to the blood bag and connection to the catheter, and these should be used in all cases.

Blood collected into syringes from cats should be transferred into a blood bag that has had the anticoagulant removed. This then enables a blood administration set to be attached in the normal way.

The rate of infusion depends on the cardiovascular status of the patient. A rate of 0.25–1 ml/kg/hr is used for the first 20 minutes. If no reactions have occurred this may be increased so that the transfusion is given with 3–4 hours. In patients who have cardiovascular or renal disease, the rate should not exceed 3 ml/kg/hr.

Monitoring the recipient

Before transfusion baseline parameters should be obtained and recorded:

- PCV;
- temperature, pulse and respiration rates;
- capillary refill time (CRT) and colour of mucous membranes.

During transfusion the patient should be monitored constantly and vigilantly. In addition, the VN should:

- monitor vital signs every 30 minutes;
- measure PCV half way through transfusion to check for haemolysis;
- monitor urine colour;
- observe for any evidence of a transfusion reaction.

Following transfusion the same parameters are recorded at 12 and 24 hours.

Clinical signs of transfusion reaction

Transfusion reactions should be rare in cases where correctly matched blood has been administered. However, it is essential to be aware of the potential risk of any transfusion, be able to recognise the signs and act appropriately. Signs include:

- discomfort, crying;
- circling, tremors;
- facial oedema;
- urticaria;
- tachypnoea or dyspnoea;
- tachycardia or bradycardia;
- vomiting;
- increased temperature;
- collapse;
- haematuria.

In the case of a transfusion reaction, stop the blood administration and contact the attending veterinary surgeon immediately. Set up and administer intravenous crystalloid therapy and administer supportive therapy as directed by the veterinary surgeon (e.g. corticosteroids, oxygen, antihistamines and adrenaline). Antihistamines and steroids are sometimes given prior to transfusion to help prevent minor reactions occurring.

Apart from mismatching of blood, other causes of transfusion reaction include:

- bacterial contamination of donor blood;
- inappropriate rate of infusion (too fast);
- inappropriate quantities of anticoagulant used;
- use of an administration set without a filter;
- old blood, or blood that has been incorrectly stored.

Chapter 42

First Aid
The A–Z of Specific Conditions

A VN may only render first aid in an emergency for the purpose of saving a life or relieving pain or suffering until the veterinary surgeon can be obtained.

ABDOMINAL RUPTURE

Abdominal ruptures may be caused by road traffic accidents (RTAs), bites, gunshot wounds and following abdominal surgery. The abdominal viscera escape through a tear in the abdominal muscle wall and may become strangulated (blood supply to escaped viscera becomes compromised or totally cut off). The abdominal viscera may or may not be protruding through the skin. Occasionally, no external wound is seen, but the escaped viscera lie under the skin surface.

(1) Wash hands and put on gloves.
(2) Clean the open wound and viscera – use warmed sterile saline to remove gross dirt.
(3) Prevent damage and further contamination of the exposed viscera.
(4) Cover viscera with a swab soaked in warm, sterile saline.
(5) Cover sterile swab with cling film or clean plastic bag to help prevent swab drying out.
(6) Apply abdominal bandage.
(7) Treat for shock and constantly monitor patient.
(8) Prevent self-trauma – Elizabethan collar.
(9) Check under bandage every 15 minutes to ensure that viscera are not being damaged by the abdominal bandage.
(10) Prepare for surgery.

ASPHYXIA (SUFFOCATION)

This can occur as a result of many conditions, such as airway foreign body, collapsed trachea, swelling of pharynx, poisoning, thoracic crushing injuries, inhalation of noxious fumes and gases, and pneumothorax. The first aid treatment of these specific conditions are covered, however, it is essential that the VN knows the clinical signs of asphyxia and general first aid treatment.

Clinical signs include:

- air hunger – gasping for air, open mouth breathing, exaggerated respiratory movements;
- orthopnoea – animal assumes position to aid respiration;
- tachypnoea;
- hyperaesthesia – these patients are excitable, stressed and react dramatically to any stimulus;
- cyanosis – often not seen until situation is extremely severe and death occurs soon unless therapeutic action is taken immediately (carbon monoxide poisoning is an exception – mucous membranes become cherry red);
- tachycardia.

General treatment.

- Do not stress.
- Remove cause of asphyxia if possible.
- Provide oxygen (see Chapter 40).
- Handle as little as possible.
- Place in quiet, dark room.
- Reassure – being unable to breathe is a terrifying experience. Calmly and gently reassure the animal.
- Treat for shock.
- Observe constantly (but this is probably one of the few occasions when you should not obtain frequent temperature and respiration rates – this just stresses the animal further).

BURNS

Common causes of burns include heat pads, electric cords, house fires, ceramic hobs, chemicals and hair dryers. The full extent of the injury may not be evident for 2–3 weeks after the initial burn has occurred; however, if the burn area is evident or known, immediate first aid treatment should be given.

(1) Apply cold water to the burn area; hose or garden spray or soaked sponges or swabs.
(2) Avoid using wet towels or ice packs if possible (creates painful pressure on the wound).
(3) Treat for shock.
(4) Keep the animal's environmental temperature warm to avoid hypothermia; use blanket and bubble wrap and avoid direct heat sources such as heat pads or heat lamps.
(5) Clip and clean the area (this may not be possible in the unanaesthetised patient in which case leave until later).
(6) Apply sterile waterproof non-adhesive dressing to prevent loss of body fluids (a clean sheet of polythene applied directly over the area with cold wet flannel is a useful temporary measure).

CHEMICAL BURNS

(1) Restrain the animal's head or apply Elizabethan collar to prevent it licking the chemical.
(2) Wash the burned area with copious volumes of water to remove the chemical from the skin.
(3) For known alkaline chemicals, e.g. caustic soda, apply an acid solution of vinegar and water in equal quantities to neutralise the chemical.
(4) For known acidic chemicals use an alkali, apply a concentrated solution of bicarbonate of soda.

CHEST WALL INJURIES

Injury to the chest wall is painful and dyspnoea, pain, collapse and subcutaneous emphysema often result. These injuries are usually caused by road traffic accidents (RTAs), dog bites, gunshot or staking injuries. Clinical signs may not be seen, however, the main complication of chest wall injuries is collapse of the lung. Damage may result in an open pneumothorax, closed pneumothorax, haemothorax or diaphragmatic rupture.

(1) Administer oxygen (see Chapter 40).
(2) Do not remove any penetrating foreign bodies.
(3) Cover wounds with 'clingfilm' and dressings to prevent further pneumothorax.
(4) Strict rest in quiet room/kennel.
(5) Prepare for radiography.

CONCUSSION

The signs of concussion vary, ranging from dazed and confused to unconscious. Other signs possible include shock, haemorrhage from ears, nose or mouth, unequally dilated pupils (anisocoria), nystagmus, slow shallow respiration, vomiting and paralysis.

(1) Maintain airway.
(2) Monitor eye reflexes and degree of depression.
(3) Treat for shock (warmth, fluids, etc.).
(4) Beware of unusual behaviour.

CONVULSIONS/SEIZURES

There are many causes that result in convulsions or seizures, including poisoning, tumours, heat stroke, epilepsy, brain trauma and metabolic disturbances (hypoglycaemia, hypocalcaemia). Most seizures result in unconsciousness and severe involuntary skeletal muscle contraction. Other signs include restlessness, salivation, pupil dilation and voiding urine and faeces.

Minor seizures may just result in change in behaviour – the animal becoming 'vacant'. It is essential that you remain calm, and advise owners to remain calm; although it looks frightening, in most cases, seizures are not fatal.

(1) Do not restrain.
(2) Move furniture away.
(3) Keep in dark and quiet room.
(4) Time length of seizure.
(5) Obtain brief history from owner (possible exposure to poisons, lactating female, RTA).

CORNEAL DAMAGE

Ulcers, chemical splashes, paint and other toxins may cause surface damage to the corneal surface. Cat fight injuries, cactus spines and RTAs may cause penetrating wounds to the cornea. Surface damage may not be noticeable but there may be a blue/opaque patch on the eye. Penetrating injuries may result in the surface of the eye looking wrinkled.

(1) Flush eye with sterile saline or water (tap water only when saline not available) to remove toxins, etc.
(2) Confine in darkened room.
(3) Do not disturb or remove any foreign bodies.
(4) Apply an eye patch/head bandage.
(5) Apply an Elizabethan collar.

DISLOCATIONS/LUXATIONS

A dislocation (or luxation) is the displacement of the articular surfaces of a joint. It commonly occurs in the carpal and tarsal joints, the hip joint and the elbow following RTAs. Clinical signs include unusual mobility, swelling around the joint, deformity, pain and limited movement of the joint.

(1) Do not reduce the dislocation.
(2) Apply cold compress to the area to reduce swelling and ease pain.
(3) Confine animal to prevent excessive movement.
(4) Analgesics as directed by the veterinary surgeon.

ELECTROCUTION

Electrocution occurs when high voltage passes through the body. It is most commonly seen in young dogs chewing electric cables. The animal is usually found collapsed by the source of the problem. Clinical signs include thermal burns (necrosis of lips and tongue), dyspnoea, muscular contractions, body stiffness and unconsciousness.

(1) Switch off electricity supply before touching the animal. If this cannot be done, move the animal away from the source with a wooden pole.
(2) Maintain airway, administer oxygen and intermittent positive pressure ventilation (IPPV) if necessary.
(3) Check heart rate – give cardiac massage if necessary.
(4) Treat thermal burns.

EPISTAXIS

Nasal haemorrhage may be caused by trauma to the head, tumours, foreign bodies and persistent sneezing. Clinical signs include haemorrhage, sneezing, open-mouth breathing, upper respiratory noise and dyspnoea.

(1) Apply cold compress to nose.
(2) Constant observation.
(3) Confine and calm patient.
(4) Sedate if necessary under veterinary surgeon direction.

FISH-HOOKS

Fish-hooks have barbed ends and will cause pain and damage to the tissues if they are pulled out. The hook should be pushed further so that the barb exits out through the skin. The shaft of the hook can then be cut with wire cutters. The hook may then be pulled out. An antiseptic such as povidone – iodine should be applied to the area. Sedation of the patient may be required for removal of the hook.

FLAIL CHEST (SEE ALSO CHEST WALL INJURIES)

Flail chest is when multiple rib fractures result in a 'floating' section of the chest. This results in poor aeration of the underlying lung.

(1) The animal should be placed into lateral recumbency with the affected side lowermost. This will stabilise the flail from moving.
(2) Provide oxygen.
(3) Manual ventilation may be required to stabilise the respiratory pattern of the patient.

FRACTURES

A fracture is a break in a bone. There are different types of fracture and they are classified as shown below.

Simple: the bone is broken cleanly, into two pieces.
Compound: the fractured bones exit out of or can be seen through a skin wound.

Complicated: where other important structures have been damaged by the fractured bone (blood vessels, spinal cord, lungs, nerves, etc.).
Comminuted: where there are many pieces of fractured bone at the fracture site.
Multiple: where the bone is fractured in more than one place or other bones are fractured.
Avulsed: where the fractured bone fragment is being pulled further away from its original position by a tendon.
Over-riding: each end of bone is pushed past the other piece.

Clinical signs seen with fractures include pain at the site of the fracture, swelling, loss of function, deformity of a part or limb, unnatural mobility and crepitus.

(1) Confine the animal to reduce movement of the fractured area.
(2) Control haemorrhage from wounds and compound fractures.
(3) Treat for shock.
(4) Clean wounds and compound fracture sites with warmed sterile saline and apply sterile dressing (refer to Wounds section).
(5) Support the fracture – however, there is some dispute about whether or not to apply splints, Robert Jones bandages, etc. Applying such support to a conscious animal could cause more pain and injury than simply putting the animal in a cage to restrict movement. Most animals will not use a limb if it is fractured and will protect it naturally themselves, when lying down or moving. Splints, Robert Jones bandages or casts can be applied if necessary, when the animal is later anaesthetised for radiography.
(6) Keep animal comfortable – provide padding and warmth.

GASTRIC DILATATION/VOLVULUS (TORSION)

This condition is most frequently seen in large, deep-chested breeds of dog and, often, shortly after they have eaten. The stomach fills up with gas and subsequently swells enormously. Sometimes the stomach then twists on itself and causes the torsion. In either case, this is a high priority emergency situation. The animal's circulatory system becomes severely compromised very quickly and the condition can be fatal if not treated immediately.

Clinical signs include pain – animal in 'praying' position (orthopnoea), restlessness, abdominal swelling, laboured respiration and collapse.

(1) Relieve pressure in stomach – pass stomach tube or if this is not possible, quickly aseptically prepare the skin to place an 18 gauge needle/intravenous catheter through the left abdominal wall at point of most distension.
(2) Administer fluid therapy to support circulation – compound sodium lactate at an infusion rate of 90 ml/kg/hr.
(3) Monitor and record vital signs.
(4) Prepare for emergency surgery – a twisted stomach will lead to ischaemic tissues and may result in removal of part of the stomach.

Note: Avoid acepromazine or other depressants, which will exacerbate the hypovolaemia.

HAEMORRHAGE

Haemorrhage may be caused by numerous types of injuries. It should be treated quickly, whether the blood loss is acute or chronic; both can result in hypovolaemia, shock and death.

(1) Apply direct digital pressure to the wound using clean hands or sterile swabs. This is useful as a temporary measure in small wounds but should not be used in the presence of a penetrating foreign body in case object is pushed further into the wound.
(2) Artery forceps may be used to close off specific arteries or veins if it is possible to locate such bleeding points. Care must be taken not to damage too much surrounding tissue.
(3) Gauze swabs or dressing pads may be applied firmly to limb wounds and bandaged into place.
(4) Doughnut ring bandages may be used if there is a penetrating foreign body.
(5) If the blood soaks through the first layer apply more layers but do not disturb clot by removing the first layers.
(6) For internal bleeding a crepe bandage may be applied to increase the back pressure.
(7) Apply a tourniquet to occlude an artery by pressing it against a bone to prevent further blood loss at a site lower down a limb or the tail.

There are three points in the dog and cat where tourniquets can be applied.

- Brachial artery: medial side of the humerus, distal third. Pressure applied prevents haemorrhage from below the elbow.
- Femoral artery: medial aspect of the thigh, proximal third. Pressure applied will stop haemorrhage from below the stifle.
- Coccygeal artery: underside of the tail, pressure is applied at the root of the tail.

How to apply a tourniquet.

- A tourniquet may be applied a few inches above the wound (between the heart and the wound).
- Adjust the tightness of the tourniquet to stop the blood flow.
- A tourniquet must not be left on more than 15 minutes before it is moved or slackened to allow the tissues to recover.
- Tourniquets should only be used when other control methods have failed.

HEATSTROKE

Heatstroke occurs from severe hyperthermia. The normal cooling mechanisms of the body are overwhelmed. Extreme temperatures (>41°C) can cause brain damage and subsequent organ failure. Usual causes are environmental. Brachycephalic breeds, animals with laryngeal paralysis and thick-coated animals are also at risk. Common clinical signs include excessive panting and increased heart rate often with hyperaemic mucous membranes. The animal requires active cooling by wetting the patient with cool water and directing cooling fans on them. Wet towels can be useful if the water shower is stressful to the patient, however they can impede the heat loss. Ice-cold water showers or baths should not be used as this could cause vasoconstriction of the vessels. The temperature should be monitored and cooling methods stopped once the temperature reaches around 39°C to prevent rebound hypothermia. In the more severe cases intravenous fluid therapy and oxygen supplementation should be implemented.

HYPOCALCAEMIA (ECLAMPSIA, LACTATION TETANY)

This is most commonly seen in lactating bitches with large litters, 2–5 weeks after parturition. The milk demand by the offspring causes huge drains on the dam's calcium levels. If the diet does not provide enough calcium the dam becomes hypocalcaemic (low blood calcium levels). Signs include (in increasing severity), restlessness, panting, tachypnoea, muscular spasms, collapse, hyperaesthesia, unconsciousness and, latterly, death.

(1) Veterinary surgeon to administer 10% calcium solution intravenously. Calcium borogluconate (0.5–1.5 ml/kg of 10% solution over 20–30 minutes).
(2) Constantly observe and monitor dam during administration (hypotension may develop after intravenous administration).
(3) Remove offspring and use an alternative feeding method for them.

HYPOGLYCAEMIA

This may occur in the diabetic patient when either too much insulin has been given, or if the animal has not eaten in the period after administration of insulin, or if the animal has overexercised. Blood glucose levels become dramatically reduced and cause signs such as lethargy, apparent blindness, ataxia, collapse, convulsions, coma and death.

(1) Give oral glucose solution, chocolate, honey, glucose (rub on to gums if the animal is unconscious).
(2) Veterinary surgeon to administer glucose by intravenous injection (1–5 ml 50% dextrose slowly over 10 minutes, ideally via a jugular catheter as 10–50% solutions are irritant).
(3) Constantly observe and monitor patient during administration.

INSECT STINGS

The mouth and front paws are the most common areas affected by wasp and bee stings. Stings may cause excessive swelling, discomfort and, in some cases, severe allergic reaction resulting in collapse. If the animal has been stung in the back of the throat, the swelling may be enough to cause serious airway obstruction.

(1) Treat for shock.
(2) Provide oxygen therapy if animal is dyspnoeic.
(3) Veterinary surgeon to give corticosteroids intravenously.
(4) Scrape the 'sting' away if it has not been embedded into the skin or pull sting out using forceps if it is already embedded into the skin. Grasp sting with forceps as close to the skin surface as possible to avoid bursting the poison sacs.
(5) If the owners know what stung the animal:
    - use bicarbonate (1 tsp bicarbonate: 250 ml water) to bathe and neutralise the area for bee stings;
    - use vinegar (50:50 with water) for wasp stings to bathe the area.

PARAPHIMOSIS

Constriction of the prepuce around an engorged penis so that the penis cannot be retracted back into the prepuce can occur in the dog. If this is neglected, gangrene of the tip of the penis may result. Clinically, the penis is very dry, swollen and reddened.

(1) Apply a cold compress.
(2) Apply KY gel or liquid paraffin to tip of penis to lubricate tissues.
(3) Reduce the paraphimosis if possible. If not possible, keep the penis moist until surgical repair is carried out.

POISONING

It is important that the VN has a basic understanding of the first aid treatment of any animal that has been poisoned, so the VN needs to know a lot of background knowledge.

Poisons enter the animal's body by:

- ingestion (eating, drinking);
- inhalation (inhaling noxious fumes);
- absorption (absorption through paws, eyes, mucous membranes, etc.).

The VN must obtain a history from the owner to aid diagnosis. Many of the clinical signs seen in poisoning cases can be confused with other illnesses and disorders, e.g. epilepsy, heatstroke, severe gastroenteritis.

Identify the type of poison

The VN should obtain a case history from owner by asking questions.

- Did you see the animal eat the poison? If so, they should be asked to bring a sample to the surgery.
- What poisons are at home? Give suggestions, often people don't even know that certain substances are poisonous.
- How long ago was it eaten?
- Has the animal been sick?

The VN should examine the animal, record temperature, pulse and respiration and act on these if abnormal. Samples should be obtained of voided body fluids (vomit, blood, urine, faeces).

Prevent further absorption of the poison

- Induce emesis if within 4 hours of ingestion, only if poison is non-corrosive.
- Administer gastric lavage.
- Administer oxygen or plenty of fresh air if the poison was inhaled.
- Flush area with copious amounts of water if poison was absorbed through skin.

Remove the poison

Emesis may be induced if the poison was ingested within the last hour: it must not be induced if a corrosive substance ingested (e.g. disinfectants – phenols, quaternary ammonium compounds, petroleum products).

Induce emesis

- Apomorphine 0.04 mg/kg I/V or 0.08 mg/kg I/M (under veterinary surgeon direction).
- Washing soda crystal (walnut-sized) (may be corrosive!).
- Salt water (2 tsp in cup of warm water). May cause hypernatraemia.
- Mustard and water (2 tsp in a cup of warm water).

It is better to ensure the animal receives treatment at the veterinary surgery rather than advising owners on making their animals sick.

Perform a gastric lavage

This is useful if carried out within 4 hours of ingestion. Place a stomach tube and holding the tube up high, pour in 5–10 ml/kg of warmed water or saline. Rotate the animal to mix the fluid in the stomach. Lower the tube into a bucket and allow fluid to be siphoned off. Repeat at least ten times. Add Fuller's earth (BCK) or charcoal during last few flushes to absorb any remaining poison:

- BCK granules (1–3 tbsp. mixed into paste with water);
- charcoal (1 g/kg) mixed with water).

Administer a cathartic substance

A cathartic is a substance that encourages the passage of the ingestate through the gastrointestinal tract. In other words, it helps to speed up the elimination of the poison through and out of the body; 40% sodium sulphate solution may be used at a dose rate of 1 g/kg

Administer a demulcent

A demulcent coats the alimentary lining with a soothing substance to protect and relieve the mucosal surface along the alimentary tract. These are especially useful when a corrosive has been ingested. There are various drugs including sucralfate; however, household ingredients may also work well, e.g. one beaten raw egg, milk, 1 tsp sugar by mouth.

Treat clinical signs and keep animal comfortable

(1) Treat for shock, seizures, dyspnoea, hypothermia and hyperthermia.
(2) Give antidote if known e.g. vitamin K (Konakion™) for warfarin (rat bait) poisoning.

Most poisons are eventually metabolised by the liver and then excreted through the kidneys. Giving intravenous fluids will help to speed up removal of the poison out of the body and to dilute the toxin reducing damage to the cells of the kidney. The veterinary surgeon may administer or add a diuretic to the fluid to increase the rate of excretion.

Notes

It is important that the VN does not get involved in any discussion with owners about malicious poisonings. The VN must never agree with, suggest or provoke conversation about this subject but remain impartial at all times.

Have the telephone number of the Veterinary Poisons Information Service (VPIS) to hand always. They provide 24-hour information service (for the UK) about poisons and the methods of treatment. The VN is only allowed to contact the VPIS with the consent of the veterinary surgeon! There is a charge for this service. The numbers are:

London 020 7635 9195
Leeds 0113 245 0530

PROLAPSED EYEBALL

Injury to the orbital area (RTAs, fights, etc.) can result in the eye prolapsing. This is more likely to occur in brachycephalic breeds with a shallow orbit and protuberant eyes. The eyeball sits out from the rest of the face, is usually

inflamed and very dry. It is important to replace the eye as quickly as possible before other damage occurs to the unprotected eyeball, and the cornea ulcerates and the optic nerve becomes permanently damaged.

(1) Lubricate the eye and keep it moist using artificial tears or sterile eye lubricant.
(2) Replace eyeball by pulling eyelids over the eye – this should only be carried out by qualified personnel.
(3) Do not push the eye back into the socket. If the method above does not work, keep the eyeball lubricated using saline-soaked swabs (advise owners to use contact lens solution or ½ tsp salt dissolved in 1 l cooled, boiled water).
(4) Apply Elizabethan collar.
(5) Treat shock.

PROLAPSED RECTUM

Severe tenesmus as a result of either diarrhoea or constipation can cause the rectum to protrude through the anal sphincter. This is most frequently seen in young puppies or kittens (and hamsters!). The length of prolapse varies, but appears as a pink/red oedematous, tubular structure, like a sausage, protruding from the anus. If left, the rectum dries out and may ulcerate.

(1) Moisten and lubricate the prolapsed tissue using warm saline to clean first and then liquid paraffin to lubricate tissues.
(2) Replace prolapse by gently pinching the end of the prolapse with the finger and thumb to encourage the tip of the prolapse to turn back in on itself.
(3) If unsuccessful, apply Elizabethan collar.
(4) Prevent further straining, by spraying prolapse with local anaesthetic agent (lidocaine) until surgical correction can be carried out.

RUPTURED DIAPHRAGM

A ruptured diaphragm is usually caused by RTAs or direct trauma. The diaphragm tears and allows the abdominal organs to pass through into the thoracic cavity, which, in turn, causes the lungs to collapse. Dyspnoea in varying degrees of severity may result. These animals often take an unusual positional stance (orthopnoea).

(1) Administer oxygen therapy.
(2) Cage rest in stress-free environment.
(3) Place animal on a sloped surface, so that head is higher than hindquarters. This encourages the abdominal organs to fall back into the abdomen and reduces pressure on the lungs and heart.
(4) Monitor patient constantly until surgery.

SHOCK

Shock occurs in nearly all accident and trauma victims. The VN must never underestimate the effects of even minor accidents on the cardiovascular system. The VN's role in providing any first aid treatment for animals is mainly concerned with controlling the physiological effects of shock. The VN must be fully familiar with the clinical signs. One should not wait until an animal is showing signs of shock, but instead prevent shock occurring in the first place. This means, that the VN should initiate basic shock control for all accident victims unless otherwise instructed by a qualified member of staff.

The clinical signs of shock become more pronounced the more severe the shock is, but include in increasing severity:

- pale, clammy mucous membranes;
- slow capillary refill time (>2 seconds);
- tachypnoea;
- tachycardia;
- rapid but feeble weak pulse (may not be able to palpate a peripheral pulse);
- cold extremities;
- hypothermia;
- depression, lethargy and collapse.

Treatment.

(1) Correct or remove the cause of shock, e.g. control haemorrhage (refer to specific first aid treatments as listed).
(2) Provide warmth, and prevent further heat loss – blankets, space blankets, etc.
(3) Prepare warmed intravenous fluids.
(4) Observe constantly.
(5) Monitor and record vital signs. It is essential that findings of vital signs are recorded so that trends can be determined – either improvements in the patient's condition or deterioration. Make notes on the general demeanour of the animal.
(6) Provide comfort and reassurance – these animals are often confused, stressed and disorientated. Handle them gently and stoke and encourage them during the recovery period.

SPINAL INJURY

Spinal injury usually occurs as a result of RTA or disc protrusion following chronic disease. Rabbits can sustain spinal fractures if restrained improperly. These patients are usually in intense pain, are unable to move or get up and cry out in pain if moved. Animals that are not paraplegic or quadriplegic often take up a strange positional stance and are ataxic.

(1) Do not move animal unless necessary.
(2) Carry on rigid board.
(3) Treat shock and keep warm.

WOUNDS

See also Chapter 19. There are many classifications of wounds depending on the area damaged and the extent of the damage. First aid treatment of wounds obviously depends on these factors but generally, first aid care consists of the procedures described below.

Closed wounds – contusions and haematomata

(1) Ice packs should be applied in the first instance to reduce the incidence of swelling.
(2) Pressure bandages can be applied to control haemorrhage and reduce swelling but should only be left on for up to 12 hours.
(3) In the later stages, hot compresses may help to reduce the pain.

Open wounds – lacerations, punctures, penetrating wounds and abrasions

(1) Remove the cause of the wound if still in place (e.g. fish-hook).
(2) Control haemorrhage if necessary.
(3) Control shock, keep animal warm.
(4) If possible, clip around the wounds to remove large amounts of fur (this may stress animal so leave until anaesthetised).
(5) Clean wound using copious amounts of sterile saline or water and very dilute solution of iodine if saline not available (warm the fluids to body temperature).
(6) Cover the wound with appropriate sterile dressing and bandage appropriately.
(7) Apply Elizabethan collar to prevent self-trauma.

REFERENCES AND FURTHER READING

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Appendix

Calculations

For:

- percentages of solutions;
- anaesthetic gas flow rates;
- fluid therapy rates;
- calorie requirements;
- radiography calculations.

CALCULATING SOLUTIONS AND DRUG DOSE RATES

Remember that:

- 1 ml of solution weighs 1 g;
- 1 g = 1000 mg;
- a 1% solution contains 1 g in 100 ml;
- a 2% solution contains 2 g in 100 ml, and so on;
- a 10% solution contains 10 mg/ml.

$$\%\ \text{solution} = (\text{weight (g)} \times 100) \div \text{volume of solution (ml)}$$
$$\text{volume of solution (ml)} = (\text{weight (g)} \times 100) \div \%\ \text{solution}$$
$$\text{weight (g)} = (\text{volume of solution (ml)} \times \%\ \text{solution}) \div 100$$

If you know that an animal needs a certain number of milligrams of a drug, but do not know how many millilitres or tablets that is, you can use this formula to work it out:

dose prescribed (mg)/dose per ml or concentration

Example

A dog needs 25 mg of a drug to be given by injection, and the drug is in a strength of 50 mg/ml:

$$25\ \text{mg} \div 50\ \text{mg} = 0.5\ \text{ml}$$

CALCULATING ANAESTHETIC GAS FLOW RATES

Tidal volume = the amount of gas passing into and out of the lungs in one breath. This is estimated at 10–15 ml/kg.
Minute volume = the volume of air inhaled or exhaled in 1 minute.

To work out flow rate, the animal's tidal volume must be calculated first.

- Tidal volume = 10–15 ml/kg.
- Cats/small dogs = 15 ml/kg.
- Medium/large dogs = 10 ml/kg.

To find out the minute volume, multiply the tidal volume by the respiration rate per minute.

Then multiply this by the circuit factor to obtain the flow rate:

- Ayres T-Piece and Bain = 2.5–3;
- Magill and Lack = 1–1.5.

The whole formula is:

Flow rate = body weight × 10–15 × respiration rate × circuit factor

Example

What would the flow rate be for a 30 kg dog with a respiration rate of 20/min?

30 (body weight) × 10 (tidal volume) × 20 (respiration rate) × 1.5 (circuit factor) = 9000 ml (9 l)

CALCULATING FLUID THERAPY DRIP RATES

The maintenance fluid requirement is the amount of fluid normally required by the patient over a certain period. This is calculated at approximately 50 ml/kg/24 hours.

Working out the number of drops per second

The drip rate must be worked out. Different giving sets administer different amounts of drops per millilitre:

- paediatric giving set = 60 drops/ml;
- general giving sets = 20 drops/ml.

Total amount in 24 hours ÷ 24 = hourly rate
hourly rate ÷ 60 = minute rate
minute rate × giving set drip factor = number of drops per minute
60 ÷ drops per minute = second rate

Example

A 15 kg dog needs 750 ml over 24 hours. A giving set is used that delivers 20 drops/ml.

$$750 \div 24 = 31.25$$

$$31.25 \div 60 = 0.52$$

$$0.52 \times 20 = 10.4$$

$$60 \div 10.4 = 5.7$$

So the giving set should be set at one drop approximately every 6 seconds.

CALCULATING CALORIE REQUIREMENTS

The formula for calculating RER is:

$$\text{RER (kcal)} = 70\ (\text{bodyweight in kg})^{0.75}$$

Alternatively, for animals weighing between 2 and 45 kg the following may be used:

$$\text{RER (kcal)} = (30 \times \text{current bodyweight}) + 70$$

This will give a reasonable estimation of the calorie requirements. This total figure is the amount of kilocalories required over 24 hours.

Example

A 25 kg dog following surgery would require:

$$(30 \times 25) + 70 = 820 \text{ kcal/day}$$

CALCULATING RADIOGRAPHIC EXPOSURES

Film–focal distance and the inverse square law

If the film–focal distance (FFD) is altered, the new exposure can be worked out using this calculation:

$$\text{new mAs} = \text{new FFD}^2 \div \text{old FFD}^2 \times \text{old mAs}$$

Example

Having been using an exposure of 10 mAs and FFD of 50 cm, what exposure is required if the FFD is changed to 100 cm?

$$10 \times 10\,000 \div 2500 = 40 \text{ mAs}$$

The 10 kV rule

- Increase the kV by 10 and you can halve the mAs.
- Decrease the kV by 10 and you can double the mAs.

If you need to keep the actual image the same, but need to alter the exposure, the 10 kV rule works like this:

50 kV at 32 mAs will create the same exposure as:
60 kV at 16 mAs and
70 kV at 8 mAs

Example

If an exposure at 20 mAs at 80 kV is adequate, what is the kV required if the mAs is changed to 10?

The mAs is halved therefore increase kV by 10 = 90 kV.

Milliamperage (mA) and time (s)

$$\text{mAs} = \text{mA} \times \text{s}$$

If mA is doubled, the time may be halved, thereby decreasing exposure time and reducing the possibility of movement blur.

Grid factor

$$\text{mAs} \times \text{grid factor (GF)} = \text{exposure required using grid}$$

Example

The exposure factors required for a lateral radiograph of a dog's abdomen are 3.2 mAs at 80 kV. If a grid with a grid factor of 3 is used, the new exposure will be:

$$3.2 \times 3 = 9.6 \text{ mAs at } 80 \text{ kV}$$

This exposure time may be too long, so the 10 kV rule can be used. By increasing the kV by 10 it is possible to halve the mAs, giving a setting of 90 kV at 4.8 mAs.

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Numbers in *italics* refer to Figures
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