Surgical Skills for Nurses

Mini Series

Session Three: Wound Closure Considerations and Post-operative Care

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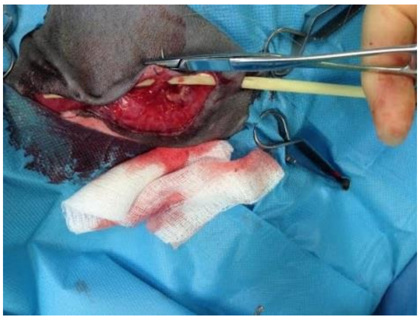

Indications for Surgical Drainage

Any drain is a temporary surgical implant providing an exit to allow removal of fluid (blood, serum, exudate), foreign material, or air from a wound or body cavity.

In wound management the correct use of a drain will improve healing, and reduce rates of infection and wound breakdown. The likelihood of wound infection is affected by a number of factors, including host factors (any physiological impairment of the animal to heal), bacterial factors (the type and number of bacteria present), and local factors (the conditions within the wound). During the wound management we can try and reduce the effect of local factors by good surgical technique. Local factors that increase infection rates include seromas, haematomas, necrotic tissue, foreign material (eg suture material, especially braided material) and dead space.

The use of a drain is indicated for two reasons:

1) To remove fluid and prevent it accumulating in a wound.

2) To prevent 'dead space'.

REMOVAL OF FLUID: the presence of fluid in a wound increases the risk of infection and slows healing. The fluid interferes with the host immune defence mechanisms towards bacteria in the wound and provides an environment for bacterial growth. The fluid may increase pressure on surrounding tissues and compromise blood supply, or prevent flaps or grafts adhering to the wound bed. Bearing this in mind, use drains where:

-Fluid will form again after needle drainage- eg seroma

-Fluid cannot be totally removed- eg abscess

-A wound cannot be completely debrided, or a source of infection persists.

-Accumulation of even a small amount of fluid will have serious consequences, eg beneath a skin flap or graft.

PREVENTION OF DEAD SPACE: dead space is an abnormal space within a wound, containing fluid or gas. Dead space is likely to occur where subcutaneous dissection is extensive, or a large mass is removed. By removing dead space we are promoting overlying tissues uniting to those below, and preventing the likelihood of the space filling with fluid to form a seroma.

Other means of reducing dead space are available, such as tacking sutures and bandaging. Tacking sutures alone are often not sufficient to reduce all dead space, they also increase the amount of foreign material implanted in a wound, this is a serious consideration in contaminated wounds. Bandaging applies pressure to keep layers apposed and reduce dead space, this may be of more use on the extremities than on the trunk, neck, or head. In most wounds, appropriate use of surgical techniques, bandaging and drains are used in conjunction as appropriate.

Types of Drain

The most important classification to make is by mode of action of the drain, ie Passive drainage, or active drainage

PASSIVE DRAINS:

Passive drains provide an exit from the drain, allowing overflow of fluid along a path of least resistance. To achieve this they need gravity on their side, so need to be placed dependently to allow flow. They will also be affected by pressure differentials within the wound. Passive drains normally function by the fluid passing over their surface, so their surface area dictates how well they work.

Obvious problems are that when placed in certain areas where there is a lot of movement, (eg axilla) they may allow air to be sucked in via a negative pressure differential. They also provide a potential route of contamination into a wound- this is combated by being dependant, the flow of fluid out of a wound, and being covered by a bandage that must be changed before any strike through.

Most common of the passive drains are Penrose drains. Other types include Yeates drains, and corrugated drains.

ACTIVE DRAINS:

An active drain has an external source of suction applied to remove fluid, or air, from a wound . Suction increases efficiency, meaning larger volumes of fluid are removed, and also allows drainage against gravity so that the drain doesn't need to be dependant, making placement easier in certain areas. As suction is applied the drain needs to be more rigid to prevent collapse of the lumen, fenestration of the portion of the drain within the wound improves efficiency, and fluid exits from the lumen of the drain rather than over the surface. As drainage is within the lumen, there is no need for a large exit wound around the drain, this combined with the suction applied, means ascending infection is much less likely, for this reason the use of active drains is indicated in clean wounds.

Active drains can be further catergorised by the type of suction applied, closed vs vented, and continuous vs intermittent, and portable or fixed.

Closed vs Vented; during closed suction, negative pressure is applied to a single lumen drain with no air vent, the incidence of infection, seroma and haematoma is much less than with active drains. In vented suction, negative pressure is applied to a multi lumen drain, ie exudates and air exits the wound as air is sucked into the wound via another lumen. The passage of large amounts of air through a wound can be traumatic and increase risk of infection.

Continuous vs Intermittant; Continuous suction is more effective at removing large volumes, and the uninterrupted flow helps prevent occlusion. Intermittent drainage is applied at least every 6 hours.

Portable vs fixed; Portable suction devices are bandaged to the animal and re-suctioned or emptied as required. Fixed suction is a wall mounted or mobile suction machine. Portable devices are much more convenient with our patients.

The most common form of suction applied to our patients is closed, continuous, portable suction. This relies on a vacuum reservoir that is attached to the animal and continuosly connected to the active drain.

Placement of Drains

Whether passive or active drains are placed, general guidelines can be applied to all drains

- Drains should be soft, radiopaque and non irritant
- Drains MUST NEVER EXIT THROUGH THE SUTURE LINE
- Place skin sutures carefully to avoid trapping the drain
- The minimum number of exit holes, and the minimum number of drains to do the job should be used.

- A large area of hair should be clipped around the exit
- The area should always be dressed
- The protruding drain and exit should be cleaned twice a day to avoid ascending infection.

Used correctly drains reduce infection rates and speed healing, but placed incorrectly, or neglected once placed, they will increase infection and cause a whole new set of complications.

Placing a Passive Drain

For passive drains the above principles need to be adhered to, but also

-For surface acting drains the exit needs to be large enough to allow fluid to exit

- The exit must be dependant

The Penrose drain is the most commonly placed, surface acting drain:

- The Penrose drain is placed prior to closure and exits through a separate dependant stab incision.
- A pair of curved artery forceps are used to create a subcutaneous tunnel from the ventral limit of the wound to the exit point.
- A stab incision is made over the point of the forceps, and the drain grasped and pulled into the wound.
- The wound is anchored dorsally using an absorbable suture placed through a small single thickness bite through the drain.
- The drain is cut so that 2cm protrudes, and anchored to the skin at the exit.

Variations include anchoring the suture dorsally with a skin suture passed blind through the drain, or exiting the drain above and below the wound- both of these increase the risk of wound infection.

Do not place passive drains in a 'clean' wound.

Placing an Active Drain

Active drains are placed prior to closure through a separate stab incision. Make sure all the fenestrated portion of the drain is within the wound to allow negative pressure to be maintained. To maintain negative pressure within the wound, the closure of both the incision and the drain exit must be airtight!

As active drains are not dependant of gravity, the exit hole does not have to be dependant. Some commercial drains have a trochar to allow placement, otherwise the drain is grasped with artery forceps as for a Penrose drain.

Active drains are not anchored in the wound, so a Chinese finger trap suture is required at the exit. To maintain negative pressure all of the wound and the exit must be airtight.

The vacuum reservoir can be a commercially produced item or improvised. Commercially available reservoirs are either rigid and must be evacuated, or more commonly are soft and compressible 'grenades' or 'concertina' types. Improvised devices can be made from either syringes with a pin placed across the plunger to maintain vacuum, or by using glass vacutainers.

The reservoir must be watched closely, and re-suctioned as soon as vacuum is lost or it fills with fluid. Whichever vacuum container is used, make sure you have a means of clamping off the drain as the reservoir is re-suctioned, this maintains the 'atmospheric bandage' effect, and prevents possible contamination being sucked into the wound if the suction reservoir is removed.

The drain itself can also be improvised from butterfly needle and attached tubing, or infusion set extensions.

Drain Removal

Ideally a drain is removed once it has served its purpose and drainage subsides, usually 3-5 days. Drains placed to eliminate dead space and promote the adhesion of tissue within a wound may need to be left longer Remember that the drain itself is a foreign body, so the presence of the drain itself will produce some fluid, this could be upto 50ml/day for an extensive wound. Remove the drain once the fluid produced is serosanguinous and of a consistant volume.

Leaving a drain in place longer than is necessary increases the risk of infection and the formation of a sinus tract. In the case of an anchored Penrose drain, after cutting the suture at the exit point, a sharp controlled downward tug is required.

Inspect the drain to ensure it is intact and all of it has been removed!

Complications

Wound Infection; the presence of a drain within a wound can affect local tissue defences to bacterial infection through pressure necrosis and foreign body reaction. The presence of an exit hole leads to risk of ascending infection. In a CLEAN wound the presence of a drain, especially a passive drain, increases infection rates. The risks are minimised by using the smallest number, of smallest diameter drains possible to do the job. Strict asepsis should be followed during placement, and during the following management. They should be left in place for the minimum necessary time.

Wound Dehiscence; As mentioned drains should not exit through the incision line, and exit holes should be kept to an adequate size only, to prevent dehiscence and herniation.

Retention or Removal; Attach drains securely and cover with a dressing, use 'Buster' collars to prevent patient interference. If fenestrations have been made too large, or too large a bite taken with the anchoring suture, it is possible for a drain to break at removal and some of it be left behind. Always inspect the drain on removal, and use radiopaque drains in case.

Failure of drainage; drains can become blocked with debris or discharge, it should only be flushed if it is placed in a contaminated or infected wound, otherwise retrograde flushing could introduce infection into the wound. Failure may also be caused by loss of suction, or improper placement- eg non-dependant placement of a passive drain.

Pain or Discomfort; the presence of a drain or its anchoring may cause discomfort to the patient. Soft flexible drains help avoid this. Cellulitis is sometimes a problem along the drain tract, leading to pain, swelling and erythema.

Summary

Select your drain based on wound factors, patient factors, availability, type of drainage and cost grounds. Active drains may cost more than Penrose drains, but may be more effective, and have a lower complication rate.

Used correctly drains are an invaluable tool, but are not a magic cure-all, they are no substitute for correct wound management, and correct post-operative care.

Close monitoring of the critical patient is essential to determine the effectiveness of any treatment, and to assess the degree of improvement in condition. Just as importantly, changes can be detected that indicate deterioration is imminent; this allows intervention to prevent a crisis before it occurs. The most useful information is provided by observing a 'trend' in the monitored vital sign, rather than a single one-off measurement. To make spotting an ongoing trend easier, a recording sheet or graph is required, with the data entered at specified intervals. How often these parameters are monitored will depend on the severity of the problem and the perceived risk of deterioration. Which parameters are to be monitored also depends on the patient; this should be decided by the clinical team and recorded in the animal's nursing plan (see Table 2.1). It is far safer to frequently re-assess a few relevant parameters than to repeatedly run a whole bulk of tests that take so much time to complete that deterioration may take longer to detect. Individual practices often have a standardized 'minimum database' of information that is gathered from emergency cases on admission. Where specific problems are suspected from physical examination, more specific monitoring can be performed and further laboratory information may be required, e.g. clotting times, slide saline autoagglutination, blood gases, lactate levels.

Organ function

Respiratory system

Regular auscultation of the chest fields should be performed to detect any change in lung sounds. Lung sounds that have become muffled may indicate worsening pleural disease; an increase in lung sounds can indicate a worsening of lung or airway pathology. Respiratory rate is useful as an indicator of respiratory disease. An increased rate could indicate a developing pneumothorax for instance, although an increase in respiratory rate (tachypnoea) can also be seen with pain, pyrexia, fear or abdominal distension.

An assessment of respiratory effort can be made by observation of the patient. Changes in posture can be indicative of increased effort: standing rather than sitting, extended neck, flared nostrils, open mouth breathing and increased abdominal movement may be seen. Respiratory function can be considered adequate if partial pressures of both carbon dioxide and oxygen are within normal limits. The method of choice to monitor this is arterial blood gas analysis. Samples are usually obtained via an arterial catheter. Blood gas analysis measures the arterial partial pressure of oxygen (PaO2) (see Figure 2.2). Pulse oximetery provides an estimate of the percentage of available haemaglobin that is carrying oxygen (oxygen saturation, SpO2) (see Figure 2.3). It does not reveal how the actual amount of oxygen is carried in the blood; this depends on the haemoglobin content. Oxygen saturation gives an idea of the efficiency of gaseous exchange from the inspired air in the alveoli into the body's tissues. Care is required with the placement of the pulse oximetery probe, if left in place for too long it tends to compress tissue and give a false reading. Conscious animals can have the probe placed on toe webs, lips or ears rather than on the tongue. Any animal with a reading of less than 95% SpO2 should receive oxygen supplementation. SpO2 values of 90% correspond to a PaO2 of 60 mmHg. Because of the nature of the oxygen saturation curve, below 60 mmHg there is a rapid drop in oxygen saturation, so aiming for an SpO2 of 95% or above gives some margin of safety.

Examples of areas that are monitored in critical patients

● Organ function:	Respiratory system; cardiovascular system; central nervous system; urinary system; gastrointestinal system
● Fluid and electrolyte balance:	Hydration status; fluids ‘in’ vs. fluids ‘out’
● Body temperature	
● Clinical pathology	Biochemistry; haematology; coagulation profile
● Pain scoring	
● Recumbency care	

Cardiovascular system

Heart rate and rhythm

Heart rate can be measured by palpation of an apex beat, palpation of a pulse or auscultation with a stethoscope. Where abnormal rhythms are detected, a continuous electrocardiogram (ECG) should be carried out, and abnormalities recorded. While an ECG is useful to investigate rhythm disturbance, it only shows electrical activity. Just because there is a waveform does not mean there is an output at that point; it is always important to check pulses at the same time as auscultating the heart.

Pulses

Pulses are commonly palpated on the femoral artery, but familiarity with palpating a metatarsal pulse is valuable. Much useful information is gathered from the rate, strength and characteristics of the palpable pulse. A pulse should be present for each heart beat/. If this is not the case, or there are variations in pulse strength, then an ECG is necessary to identify rhythm disturbances. Pulse rate and character are essential in detecting hypovolaemia and the response to treatment. Increasing pulse rate and decreasing amplitude are evidence of worsening hypovolaemia. The distal metatarsal pulse becomes non-palpable with moderate hypovolaemia, but should return if effective therapy is instituted. Importantly, what is palpated as the pulse amplitude is the difference between diastolic and systolic pressures (i.e. an animal with a systolic pressure of 100 mmHg and a diastolic pressure of 60 mmHg would have a similar pulse amplitude to an animal with 70/30 mmHg blood pressure); it cannot accurately measure actual blood pressure. Therefore the pulse needs to be considered in conjunction with measures of tissue perfusion and blood pressure readings.

Mucous membranes and capillary refill time

The mucous membrane colour (see Table 2.2) and capillary refill time (CRT) can help to give an idea of tissue perfusion and vasomotor tone. The oral mucosa is normally used as it is easiest to access. CRT tends to vary with an individual’s technique. A normal CRT is usually 1–1.75 s. A slower CRT suggests reduced blood flow in the tissue, often resulting from vasoconstriction with hypovolaemia, or heart failure. A more rapid CRT suggests increased blood present in the tissues; this may be due to vasodilation seen in sepsis. Mucous membranes are normally pink, although healthy cats often have paler membranes than dogs.

Observed changes in mucous membrane colour

Colour observed	Possible cause
Pale, white or grey	Poor perfusion, or anaemia
'Brick red' or 'injected'	Vasodilation, systemic, inflammatory response
Blue or purple	Cyanosis: low oxygen saturation of haemoglobin
Yellow	Increased blood bilirubin levels
Brown	Formation of methaemoglobin, e.g. paracetamol poisoning
Cherry red	Carbon monoxide poisoning

Tissue perfusion

The sole aim of the cardiovascular system is to deliver oxygenated blood to the tissues of the body. All tissues need a supply of oxygenated blood. Monitoring assesses the delivery of this blood to the capillary beds of the tissues. A range of parameters can help to form an overall picture of perfusion:

1) Mucous membrane colour

2) Capillary refill time

3) Peripheral pulse

4) Toe web temperature vs. core temperature

5) Urine output (1.0 ml/kg/hour)

6) Blood lactate levels

7) Arterial pressure.

A systolic arterial pressure of 90 mmHg (equivalent of 60–70 mmHg mean arterial blood pressure) is required for adequate flow to vital organs. It is most practical to use a Doppler system and cuff (non-invasive, indirect measurement). Alternatives include invasive, direct measurement via an arterial catheter.

Central venous pressure

In cases that require fluid therapy, but there is a risk of fluid 'overload' if too much fluid is administered, it is useful to measure central venous pressure. This gives an idea of venous 'filling' and how much fluid is returning to the heart. Examples of typical cases would be anuric/oliguric renal failure, or animals in heart failure. A central catheter is required, and the pressure reading can be taken using a manometer, or the central catheter can be connected to a pressure transducer and the wave form constantly monitored.

Central nervous system

An animal may have altered mentation because of conditions inside the skull, such as brain injury, or due to more global conditions such as hypovolaemia, hypoglycaemia or development of a systemic inflammatory response. By monitoring neurological status, and recording findings, it is possible to spot trends quickly that will highlight any deterioration or improvement in the patient's condition. The use of a scoring system allows an accurate record to be kept of the animal's status. While there is still some subjectivity involved, allocating an overall score allows a trend to be spotted, and provides continuity from one team member to another. The Small Animal Coma (SAC) scoring system tends to be used (see Chapter 14). This is an adaptation of the Glasgow Coma Score (GCS) that is used in human medicine. In the SAC system, a score is allocated from 1 to 6 for each of 'motor activity', 'brainstem function' and 'level of consciousness', giving a maximum score of 18.

Urinary system

Monitoring urine specific gravity and output allows quick and simple assessment of kidney function. Normal urine output is considered to be 1–2 ml/kg/hour. If urine output is at or above this level it is assumed renal perfusion is adequate, and therefore it is likely that perfusion of other organs is also adequate. In animals with an indwelling urinary catheter, a closed collection system provides a means of measuring urine volume. In other animals litter or bedding can be weighed to estimate the urine of volume expelled.

Gastrointestinal system

Patients that are systemically ill can develop vomiting, diarrhoea or ileus. Any vomiting should be recorded, along with any defecation and its nature. Vomiting and diarrhoea will lead to alterations in fluid requirement. The animal's appetite should be recorded, and food consumed accurately recorded to ensure sufficient requirements. Ileus can be assessed by auscultating the abdomen for the presence of gut sounds.

Fluid balance

All animals receiving fluid therapy need ongoing monitoring to assess effectiveness of therapy, and to prevent under or over-dosing. Important consideration must be given to determining what the patient's fluid needs are; is the patient hypovolaemic, dehydrated, or both? Hypovolaemic animals need rapid fluid administration, whereas dehydrated animals required correction of their fluid deficit over 24 hours. Physical assessment of perfusion parameters and hydration parameters should be carried out frequently. Patients receiving intravenous fluid therapy need to have their fluid input compared with their fluid output. Fluid input is easily measured by recording the number of fluid bags administered, or more accurately with an infusion pump. Other inputs to consider are any oral fluids or food, and intravenous drugs. Fluid output includes urine, faeces, vomit and any effusions. Urine output can be measured via a urinary catheter, or in animals that are not catheterised disposable bedding can be weighed before and after urination to estimate volume (1 gram = 1 ml urine). Cat litter trays can be weighed in the same way. Volumes of vomit and faeces can be estimated. Volume of effusions can be more difficult to determine, but outputs from thoracic and abdominal drains are easily recorded, as are wound effusions collected in active suction drains. Dressings can be weighed to estimate effusions in situations such as burns, or open abdominal drainage. Some fluid outputs are not measurable, e.g. loss of water as vapour in expired breath; these losses are termed 'insensible' losses, and are usually estimated at 20 ml/kg/24 hours. Once the fluid inputs and outputs have been established, they can be compared. Any large discrepancies should be investigated. In a hypovolaemic patient we would expect the 'ins' to be much greater than the 'outs' as the deficit is corrected. In a patient with normovolaemia, the 'ins' should be slightly greater than the 'outs'.

Patients should also be weighed accurately at least twice a day; any large gains or losses are likely to be caused by fluid imbalance.

Body temperature

Prolonged abnormal body temperatures can cause potentially fatal organ dysfunction. Abnormal body temperatures interfere with a patient's homeostatic mechanisms, and so delay return to normal health. Critically ill animals are less able to regulate their body temperature. Where active warming is employed in hypothermic animals, care must be taken not to cause overheating, or localized burning.

Clinical pathology

Blood glucose

As well as the obvious cases where blood glucose levels are important, such as monitoring a diabetic ketoacidosis patient, control of blood glucose levels are essential in other critical patients. Hypoglycaemia is commonly seen in hypovolaemia, sepsis, hyperthermia and liver disease. The use of handheld glucometers makes glucose level testing quick and easy, and allows rapid adjustment of glucose supplementation via intravenous fluids.

Packed cell volume and total protein

Trends in packed cell volume (PCV) and total protein (TP) can be interpreted together to give information regarding fluid balance or ongoing haemorrhage. Changes in both may be in the same direction, but alterations in the ratio give extra information:

- Increase in PCV and TP: dehydration
- Decrease in PCV and TP: aggressive intravenous fluid therapy (IVFT), haemorrhage (later, after interstitial fluid moves into intravascular space, initially no change or even increased PCV with decreased TP, due to splenic contraction)
- Decreased PCV, normal TP: increased destruction of red blood cells?
- Increased PCV, decreased TP: dehydration with protein loss, e.g. haemorrhagic gastroenteritis (HE).

PCV and TP are important in guiding fluid therapy and choice of fluid, e.g. colloid, crystalloid.

Electrolytes

Electrolyte disturbances are common in critical patients, either because of their presenting complaint or as a result of fluid therapy or drug administration. Electrolytes should be repeatedly checked during hospitalisation. Most isotonic crystalloids used for fluid maintenance (e.g. Hartmann's solution) have insufficient potassium levels for maintenance requirements.

• Basic nursing monitoring	Advanced nursing monitoring
Heart rate, pulse rate and quality, capillary refill time, mucous membrane colour every 2–12 hours	Blood pressure measurement continuous or every 2–12 hours
Respiratory rate and effort, auscultation of lungs every 2–12 hours	Central venous pressure monitoring every 2–6 hours
Rectal temperature every 4–12 hours	Continuous or intermittent ECG; note dysrhythmias
Measure or note urine output, or palpate bladder every 2–6 hours	Pulse oximetry continuous or every 2–12 hours
Mentation/Small Animal Coma Score every 2–6 hours (unless specific case, e.g. head trauma)	End-tidal capnography continuous or every 2–12 hours
Note any regurgitation, vomiting or faeces production every 2–6 hours	Arterial blood gas analysis every 2–24 hours
Assess adequacy of analgesia every 2–4 hours	Electrolyte measurement every 4–24 hours
Walk ambulatory patients, or turn recumbent patients every 4 hours	Nebulise and coupage for 10–20 minutes every 4–6 hours
Lubricate eyes if patient sedated/unable to blink every 2–4 hours	Check, clean and suction tracheostomy tube every 2–4 hours
Check oxygen supplementation requirements, if necessary, every 2–4 hours	Aspirate chest drains every 2–4 hours, record volume of air and/or fluid removed
Check intravenous fluid type and rate every 2 hours	Record mechanical ventilator settings, airway pressure and tidal volume every 2 hours
Check patency of intravenous catheters and flush with heparinised saline every 4–6 hours	Peritoneal dialysis: infuse dialysate, dwell and drain every 1–2 hours, record volumes and quality of fluid obtained
Check bandages for position, tightness and cleanliness every 4–6 hours, replace if necessary	
Offer food and water (specify type and calculate RER) and record volume ingested as directed/dictated by method of feeding	

Fluid balance

Many critical patients have an altered fluid balance. Depending on the individual case, they often receive large volumes of crystalloids, colloids or blood products. It is vital to measure fluids in and out over a period of time and the veterinary nurse should understand the period of time that these products are likely to be present in the patient's circulation before moving into the interstitium, being broken down in the kidneys and passed out in the urine. Fluid 'ins' can include water consumption, nutrition and parenteral fluids; fluid 'outs' include urine, faeces, vomit, wound drainage, third space losses. Patients should be weighed twice daily as an assessment of fluid loss and gain, and this information can be used in combination with clinical examination and PCV/TP to estimate fluid balance. See Chapter 2 for more information on fluid therapy and fluid balance.

Nutritional status

The nutritional status of the patient is a major consideration that should be addressed on a daily basis. Body weight, body condition score and disease processes should all be taken into consideration when assessing the patient's nutritional requirements. The patient's daily RER should be calculated and an estimate made as to whether the patient is voluntarily consuming the requirement. If not, then techniques to achieve this should be considered, including appetite stimulants or assisted feeding techniques such as feeding tube placement should be employed. Ensuring adequate nutrition is vital in the critical patient and should be initiated as early as possible.

Care of indwelling catheters and tubes

Commonly, the critical patient will have numerous indwelling tubes and drains, and it is vital that these devices are correctly managed and cared for. Patients hospitalised within the ICU are at highest risk of developing hospital-acquired infections, because of the presence of indwelling devices and decreased immune systems so correct protocols for their care are vital. When handing devices, asepsis should always be maintained, patency must be maintained to ensure catheters, drains, feeding tubes, etc. can function correctly, and complications must be kept to a minimum. Additionally, bandages should be changed twice daily as a minimum and the insertion or stoma sites checked for signs of redness, swelling, infection, etc. It is also advisable to label these bandages so incorrect medications are not administered into tubes or catheters. Aseptic technique should be strictly adhered to whenever dealing with indwelling devices. Ideally, sterile gloves, but certainly examination gloves should be worn throughout catheter and drain care procedures.

Intravenous catheters

Intravenous catheters (both peripheral and central) should be placed aseptically, flushed every 4–6 hours, the dressing removed, the insertion site checked for extravasation of fluid and infection, and dressing replaced once or twice daily. Once the catheter is no longer required it should be removed. If venous access is required at a later time, another catheter can be placed. The date of catheter insertion should be noted on the patient's records and/or hospital sheet, and administration sets should be replaced every time a new catheter is placed. Connectors and injection ports should be swabbed with alcohol and allowed to dry before being used. Infections can be minimised by keeping intravenous fluid lines as closed systems and keeping disconnections to a minimum.

Arterial blood pressure

Since cardiac output cannot easily be measured clinically, other methods of estimating blood flow to the tissues must be used. Blood pressure provides the driving force for tissue perfusion.

Blood pressure = cardiac output X systemic vascular resistance

Cardiac output = Heart rate X Stroke volume

What these mean is that blood pressure is dependent on cardiac output and systemic vascular resistance (Vasodilation/vasoconstriction). Cardiac output is dependent on heart rate and stroke volume (the volume of blood pumped out in one contraction). If either one of these decreases then the cardiac output may decrease. In healthy patients there are compensatory mechanisms that prevent a fall in blood pressure, so for example if the cardiac output falls then systemic vascular resistance will increase (vasoconstriction) to compensate and maintain blood pressure. However in sick patients these compensatory mechanisms may not be able to function normally. When the mean arterial blood pressure (MABP) is maintained in the range of about 60 to 120 mmHg, blood flow to the brain and kidney is autoregulated via local mechanisms that maintain adequate perfusion. When MABP falls below about 60 mmHg, tissue perfusion is reduced. Hypotension can result in lactic acidosis and hypoxia due to inadequate tissue perfusion. The kidneys and brain are especially susceptible to this ischaemic damage. Blood pressure can also be used as a guide to depth of anaesthesia with increasing blood pressure potentially indicating lightening anaesthetic depth and decreasing blood pressure deepening anaesthesia. If hypotension occurs, depth of anaesthesia should be decreased and IV fluids administered to effect. The use of drugs which cause vasoconstriction such as medetomidine will give a high blood pressure reading although tissue perfusion may not be adequate. There are two ways to monitor blood pressure; Direct (invasive) and indirect (non-invasive).

Invasive (Direct) blood pressure

Is the most accurate way of monitoring blood pressure but due to the cost, technical difficulty and risks, it is rarely performed in general practice. If the skill and equipment exist in your practice it may be useful for critically ill patients. A catheter must be placed directly into the artery (normally the dorsal metatarsal) which is then connected to a calibrated electrical transducer via saline filled non compressible tubing. This allows the arterial pressure waveform to be displayed on the screen. It provides continuous, real time measurements of the systolic, diastolic and mean blood pressure and displays the waveform which can provide useful information about the cardiovascular system. The catheter must be placed aseptically, flushed frequently to prevent thrombus formation, and there is a risk of infection and haemorrhage/haematoma formation.

Non-invasive blood pressure monitoring:

Oscillometric technique

This is an automated technique found in multiparameter monitoring machines, where a cuff is placed over an artery. The cuff is inflated to a pressure at which arterial blood flow is occluded, and then gradually deflated. Pressure within the cuff oscillates when the blood flow resumes and this is detected by the machine. Systolic, mean and diastolic pressures are measured. The machine can be programmed to cycle at regular intervals. The oscillometric technique may be inaccurate on smaller patients, in the presence of vasoconstriction, tachycardia, bradycardia, hypotension, hypertension and arrhythmias. In these cases the use of a Doppler probe may be more useful.

Doppler technique (Doppler ultrasonic flow detection)

The Doppler technique measures systolic blood pressure and involves the use of piezoelectric crystals which transmit and receive sound. The technique relies on the fact that the frequency of sound reflected from moving tissues (arterial blood) differs from that transmitted from the crystal. Many people find it useful as it provides an audible pulse and will still give a systolic blood pressure in cases where the oscillometric technique normally won't (bradycardia, arrhythmias and hypo/hypertension). It is certainly recommend to place a doppler probe and cuff when anaesthetising critically ill patients if you have one in your practice.

Whichever type of non-invasive blood pressure technique you use, the selection of a correct size cuff and correct cuff placement is important to ensure accurate readings. The width of the cuff should be 40% of the circumference of the limb and the bladder of the cuff should be placed over the artery. The cuff should be wrapped around the limb firmly, neither too tight nor too loose, and tape should not be wrapped all the way round the cuff as this will prevent the cuff from inflating properly. The cuff should ideally be placed at heart level. If the cuff is too loose, lower than heart level or too small, the reading will overestimate the true blood pressure. If the cuff is too tight, too big or higher than the heart, then the blood pressure machine will underestimate the real blood pressure.

Chest drains

Chest drains should be aspirated every 4–6 hours. Aseptic technique should be used whenever tubes are handled, particularly for aspiration, and when handling connection points, e.g. three-way taps. If a dressing was applied it should be removed ideally twice, but certainly once daily, and stoma sites inspected for signs of infection. The area around the stoma should be gently cleaned using a dilute chlorhexidine or povidone–iodine solution. Previously it was recommended that antibiotic cream should be used around the stoma site, but this is no longer recommended because of the incidence of multi-drug resistance becoming increasingly common, including resistance to topically applied antibiotics, e.g. mupirocin. Instead, topical anti-microbial dressings, e.g. honey, silver, polyhexamethylene biguanide (PHMB) are recommended.

Tracheostomy tubes

Twenty-four hour care is essential in patients with tracheostomy tubes in place as potentially fatal occlusion of the tube by exudate, mucus, bedding or skin folds may occur, as well as tube dislodgement. If present, the inner cannula of the tube should be removed for cleaning whenever an increased noise or effort in respiration is detected, or initially every 2 hours post-placement. The cannula should be cleaned thoroughly using warm water, allowed to air dry and then replaced. For tracheostomy tubes without an inner cannula, the entire tube should be removed for cleaning. Ideally, a spare sterile tracheostomy tube should be available for immediate replacement into the trachea following the removal of the dirty tube. The stay sutures above and below the tracheal incision should be used to gently bring the trachea to the level of the skin and to open the trachea.

Humidification: If the inner cannula or lumen of the tracheostomy tube is repeatedly full of exudate or mucous, then either nebulised air should be used for periods for the animal to inhale, or 0.1 mg/kg sterile saline should be instilled into the tube every 2 hours (the instillation of the tube may initiate transient coughing).

Suction: Suction of the tracheostomy tube should be performed only as required. It is required more frequently in smaller dogs and cats. The patient should be pre-oxygenated for 30–60 seconds before suction is performed. A sterile suction catheter should be introduced aseptically into the tracheostomy tube and suction applied for no more than 10 seconds, whilst gently rotating the suction tube. The suction catheter should remain within the tube during suctioning and only inserted into the delicate trachea if absolutely necessary to clear an obstruction distal to the tracheostomy tube.

The tracheostomy stoma should be inspected at least once, preferably twice, daily. The area should be gently cleaned using sterile saline-soaked swabs. If the above measures do not relieve breathing difficulties then the entire tube should be changed. It is important that a veterinary surgeon is on-hand and the ability to perform endotracheal intubation and oxygen administration are readily available. The patient should be pre-oxygenated and the trachea stabilised using the stay sutures around the tracheal rings, above and below the tracheostomy site. The existing tube should be removed and a new tube rapidly inserted.

Feeding tubes

For naso-oesophogeal and nasogastric tubes, the patient's nares should be cleaned using damp cotton wool twice daily. Oesophogostomy and gastrotomy tubes should be inspected and have the stoma site cleaned using chlorhexidine or povidone–iodine solution at least once, preferably twice daily. As with intravenous catheters, antibiotic cream should no longer be applied around the stoma site and a topical antimicrobial dressing applied instead. All tubes should be flushed before feeding using 5–10 ml lukewarm water. Gastrostomy tubes should have the contents of the stomach aspirated before feeding. If there is a delay in gastric emptying, and there is more than half the previous feed in the stomach, then the veterinary surgeon should be informed and the meal skipped. In this situation motility modifiers may be considered. The tube should be flushed again post-feeding using 5–10 ml lukewarm water, to maintain a column of water within the tube between feeds to minimise blockages. If tubes do become blocked they may be unblocked by using carbonated drinks, pineapple or cranberry juice to clear the blockage.

Urinary catheters

Urinary catheters must be placed aseptically and maintained as hygienically as possible to try to minimise the incidence of infection. Before placement the prepuce or vulva should be flushed with a dilute chlorhexidine or povidone–iodine solution, and then the prepuce or vulva should be cleaned in the same manner on a daily basis. The catheter should be handled aseptically and disconnections kept to a minimum to reduce the risk of contamination. All connection sites should be wiped using alcohol, and allow to dry, before and after handling. The external catheter should be wiped in antiseptic solution four times daily and the catheter removed as soon as it is no longer required to minimise the risk of infection. Collection systems should be below the level of the patient at all times (see Figure 19.5).

Nursing the recumbent patient

There are numerous reasons why a patient may be recumbent and the nursing care of these cases can be challenging for several reasons. Maintaining proper body position and care with patients who are recumbent for prolonged periods is very important. Limbs and joints should be maintained in a neutral position to avoid loss of muscle fibres which will be lost at a higher rate in a shortened position. Providing padding under and in between joints is important to maintain air circulation to prevent moisture build up and pressure sores. Patient positioning is an important consideration as it is vital to maximize oxygen exchange. The patient's body position should be alternated between left lateral, right lateral and sternal recumbency. This change in position should be carried out at least every 4 hours, ideally every 2 hours, and this position changing schedule should be part of the patient's hospitalisation chart. This change in position is important to decrease the secretions that will build up in the dependent (lowest) lung field and will also help to reduce the incidence of atelectasis. If this change in position is not carried out, and sometimes despite it being carried out, patients may develop ventilation–perfusion (V/Q) mismatch. V/Q mismatch occurs when the volume of circulating blood in the dependent lung field increases, but the lung is unable to expand fully, therefore decreasing the ability to deliver well oxygenated blood to the body. If the patient has a severely compromised lung field, they may be unable to tolerate a particular recumbency for extended periods of time. Close monitoring of the patient is vital, observing for increased respiratory rate and effort, and should this occur they should be returned to a position that allows optimum gas exchange. The use of pulse oximetry is particularly useful in these patients.

Decubitus ulcer (pressure sore) formation is another concern in the recumbent patient. Patients who lie or sit in one position for prolonged periods of time are at risk of developing decubitus ulcers. Their early appearance is erythema, oedema and tenderness, which is followed by serum exudation and hair loss. Ischaemic necrosis results in rapid loss of the skin and subcutaneous tissue. Patients should be assessed daily for any signs of ulcer formation.

Patients with decubitus ulcers are at risk of bacteraemia, with the potential risk of infecting surgical wounds or surgical implants. Prevention of decubitus ulceration is much easier than its treatment. As written above, the patient should change position frequently (ideally every 2 hours), pressure relieving bedding should be provided, e.g. plastic covered mattress with at least one layer of soft bedding, e.g. Vetbed™; air or water beds can also be useful to reduce the incidence of decubitus ulcer formation. It is vital that patients have sufficient bedding between them and a hard surface. The bedding should be checked regularly to ensure it is clean and dry, particularly in non-ambulatory patients who do not have an indwelling catheter. Vetbed™ is ideal for these patients as urine is wicked away from the patient to minimise urine scalding.

Bladder care is another important consideration in these patients. Some patients may have indwelling urinary catheters placed, which are attached to a closed collection system. The drainage bag on the system must be emptied regularly. In noncatheterised patients, manual expression may be attempted, but it is also important to ensure patients are consciously urinating and that urine overflow is not occurring. This can be assessed by palpating the abdomen to check the size of the bladder following urination. In these patients the application of a barrier film or cream (e.g. Cavilon™) to clean, dry skin is useful as it will help prevent urinary and also faecal scalding. Faecal care is another consideration for the recumbent patient. These patients may have decreased gastrointestinal motility, and they can quickly become constipated. Hospitalisation charts should note when faeces has been passed and if necessary enemas and laxative administration may be required.

It is important to perform standing or assisted standing exercises on recumbent patients to improve circulation, neuromuscular strength and proprioception.

Eye and oral care

Eye and oral care is important in all patients but more so in the recumbent patient. The mouth should be cleaned using mild oral antiseptic solution (e.g. diluted Hexarinse™) to reduce the bacterial load and reduce the incidence of bacterial translocation to the chest and risk of pneumonia. The eyes should be cleaned regularly with sterile saline and a suitable eye lubricant applied. Many patients have reduced tear production or reduced blinking, and so are at risk of ocular ulceration.

Coupage

Coupage is a useful technique for both the recumbent and also the respiratory patient. It is an extremely effective technique that utilises the percussive force of the hand to stimulate a cough response. It is performed by cupping the hands and placing them on both sides of the patient’s chest. Begin at the caudal aspect of the lung fields and move cranially, gently striking the chest wall in a rhythmic fashion. Technique is much more important than force and should be done at the level of the patient’s comfort. Generally, this technique is most productive after standing of walking exercises. Vibration is a valuable technique used to assist the patient in mobilising secretions from the lower, smaller airways into the larger airways to be coughed out. Vibration should be performed while the patient is in lateral recumbency. With locked arms vibrate the chest wall with the hands as the patient exhales. Perform vibration during 4–6 consecutive breaths. Percussion and vibration are contraindicated in patients with rib fractures, chest tubes, platelet count below 30,000, over open wounds, or in animals with severe chest pain or arrhythmias.

Embolus formation

A potentially life-threatening complication that critical patients can develop is embolus formation. Patients who are immobilised due to their critical status are at a higher risk of thromboembolism. This is due to blood pooling in dependent regions creating erratic sedentary flow, circulation compromise due to injury and activation of clotting factors due to inflammation.

Rehabilitation and physiotherapy techniques can help decrease the need for heparin therapy or thrombus formation during disseminated intravascular coagulation.

Techniques such as standing or assisted standing, massage and passive range of motion are all extremely beneficial in resolving oedema and reestablishing more normal blood flow.

Physiotherapy

Rehabilitation in the intensive care setting may improve the animal's quality of life and reduce the complications associated with prolonged hospitalisation. The requirements should be determined on an individual patient basis. The basic therapeutic modalities of massage, passive and active range of motion exercises, postural drainage, therapeutic exercise, therapeutic ultrasound and electrical stimulation can be used to decrease pain, improve function, maintain muscle tone, improve skin perfusion and reduce complications (e.g. oedema and decubitus ulcer formation).

Pain management

Pain scoring should be performed to allow an objective assessment of the patient. This is particularly important when several members of staff may be caring for the same patient. Nurses should be encouraged to communicate to the veterinarian in charge of the animal if they think the analgesia plan is not correct, and the hospital charts should have provision to ensure regular pain scores are performed.

Mental well-being

Another, often overlooked, consideration is that of the patient's mental well-being. Time should be set aside for TLC and also to allow visits by the owners. In the authors' hospital, pre-determined visiting hours are built into the day and time slots are allocated to the owners. If permitted, owners are able to feed and groom their animals and the time is taken to demonstrate physiotherapy and assisted feeding techniques to the owners. Occasionally, a patient is distressed by the owners visits when they leave, and so in this situation the visits may be discouraged. Rest is also important in the patient, particularly in busy 24-hour hospitals. Lighting, barking, infusion pump alarms and the general environment can be noisy and so where possible a period of time should be provided whereby lights are dimmed to allow the patients to rest. The timing of medications and feeds should also be taken into consideration to allow patients time to rest.