BSAVA Guide to Procedures in Small Animal Practice

Nick Bexfield and Karla Lee

The BSAVA Guide to Procedures in Small Animal Practice provides practical, step-by-step guidance on how to perform the diagnostic and therapeutic procedures commonly performed in small animal veterinary practice. In addition, routine clinical examination of the major body systems, and protocols for the management of selected emergencies, are described.

In addition to the actual technique, each procedure has information on indications and contraindications, equipment required, and potential complications, together with the editors' own hints and tips. Details of BSAVA Manuals where wider information can be found, such as interpretation of results, are given throughout.

Special features:
- A to Z format to aid information retrieval
- Extensive cross-referencing in highlighted text
- Specially commissioned drawings
- Lay-flat binding

This is a truly useful guide, which will provide a valuable and lasting reference for veterinary surgeons, veterinary nurses and students alike.

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List of abbreviations

Procedures A to Z

A
Abdominocentesis; ACTH response test; Anaphylaxis – emergency treatment; Arthrocentesis; Aseptic preparation

B
Barium contrast media; Barium studies of the gastrointestinal tract; Blood pressure measurement; Blood sampling; Blood smear preparation; Blood transfusion; Bone biopsy – needle; Bone marrow aspiration; Bronchoalveloar lavage; Bronchoscopy; Buccal mucosal bleeding time

C
Cardiopulmonary–cerebral resuscitation; Cardiorespiratory examination; Cast application; Cerebrospinal fluid sampling; Cranial draw test; Cystocentesis

D
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E
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F
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Thoracocentesis – needle; Thoracostomy tube placement; Tibial compression test; Tissue biopsy – needle core; Tracheostomy; Transtracheal wash

Urethral catheterization; Urethral retrograde urohydropulsion; Urinalysis

Velpeau sling

Water deprivation test; Whole blood clotting time
Foreword

It is with tremendous pleasure that I am able to introduce to you the first edition of the *BSAVA Guide to Procedures in Small Animal Practice*. Having read through the new title, I have absolutely no doubt this is going to be a book of huge and practical value to veterinary surgeons and their teams, up and down the country. The concept is simple. A ‘real world’ guide that provides rapid access to useable information on common practical techniques. And crucially, it is designed in a manner that ensures the information can be accessed easily and quickly in the busy and demanding environment of small animal practice. Whether you want a quick refresher on how to perform that abdominal tap, or a reminder of the protocols for endocrine diagnostic testing, the *BSAVA Guide to Procedures in Small Animal Practice* is simply designed to enable you to find exactly what you want, when you want it. Fast. Accurate. Dependable.

The team behind this new and exciting guide have worked tremendously hard to ensure that it meets the needs of veterinary surgeons with busy schedules but who need to be able to absolutely rely on the information in front of them. They have done a truly superb job and I am sure you will be as impressed with the guide as I am. Nick Bexfield and Karla Lee, the Editors of this inaugural edition should be enormously proud of the new guide. Likewise the Publishing team at BSAVA have done a great job and I would like to congratulate them all.

I am sure the *BSAVA Guide to Procedures in Small Animal Practice* will become a ‘must have’ in every good small animal practice and indeed in every BSAVA members’ coat pocket! And of course being designed exclusively for our members, it is yet another trusted benefit of membership of the BSAVA.

Richard Dixon BVMS PhD CertVR MRCVS
BSAVA President 2009–2010
Preface

It is our great pleasure to introduce the first edition of the BSAVA Guide to Procedures in Small Animal Practice. Our primary aim was to create a book dedicated to the diagnostic and therapeutic procedures performed routinely in small animal veterinary practice, with the objective of providing practical, step-by-step guidance on how to perform these techniques. In addition, this guide also includes sections on clinical examination of the major body systems, and protocols for the management of selected emergencies, including anaphylaxis and seizures.

Most of the procedures contained within this guide have originated from the BSAVA library of publications. However, they have been adapted to focus on the technical aspects of the procedure and to include hints and tips that we ourselves find useful. Each procedure also includes indications and contraindications, an equipment list, and potential complications. Procedures for sample collection additionally contain brief sections on sample handling. Photographs and other illustrations are included to provide clarification as necessary.

The BSAVA Guide to Procedures in Small Animal Practice focuses on procedural technique and therefore should be used in conjunction with other sources of information, such as for diagnostic test result interpretation. We have included throughout details of the relevant BSAVA Manuals where further information can be found.

We are grateful to the authors of the BSAVA publications from which much of the material from within this guide has been derived and also to our teachers and colleagues, who have been the source of many of the hints and tips. We must also thank the Publishing team at BSAVA for their editorial and administrative assistance. Marion Jowett deserves a special mention for her enthusiasm, patience and attention to detail. We are also grateful to Samantha Elmhurst for her careful illustrations.

We hope to have created a truly useful guide which will provide a valuable and lasting reference for veterinary surgeons, veterinary nurses and students.

Nick Bexfield
Karla Lee

December 2009
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List of abbreviations

ACD     Acid citrate dextrose
ACTH    Adrenocorticotrophic hormone
BAL     Bronchoalveolar lavage
BMBT    Buccal mucosal bleeding time
CCL     Cranial cruciate ligament
CNS     Central nervous system
CPD     Citrate phosphate dextrose
CPDA    Citrate phosphate dextrose adenine
CPCR    Cardiopulmonary–cerebral resuscitation
CSF     Cerebrospinal fluid
CT      Computed tomography
DPL     Diagnostic peritoneal lavage
DV      Dorsoventral
ECG     Electrocardiography
EDTA    Ethylene diamine tetra-acetic acid
ET      Endotracheal
GI      Gastrointestinal
HAC     Hyperadrenocorticism
i.m.    Intramuscular
i.v.    Intravenous
LMN     Lower motor neuron
MRI     Magnetic resonance imaging
PCV     Packed cell volume
RER     Resting energy requirement
UMN     Upper motor neuron
VD      Ventrodorsal
Abdominocentesis

Indications/Use
- To obtain abdominal fluid for diagnostics in cases of abdominal effusion

Contraindications
- Coagulopathy
- Marked distension of an abdominal viscus
- Severe organomegaly

Equipment
- As required for *Aseptic preparation – (a) non-surgical procedures*
- Hypodermic needles:
  - Dogs: 21 G; 1 to 1.5 inch
  - Cats: 23 G; ¾ inch
- 5 ml syringe
- EDTA, plain and sterile collection tubes
- Microscope slides

Patient preparation and positioning
- Sedation may be required.
- The patient should be restrained in right lateral recumbency.
- It may be necessary to empty the patient’s bladder by manual expression or *urethral catheterization* to reduce the risk of accidental cystocentesis.
- *Aseptic preparation – (a) non-surgical procedures* is carried out on an area approximately 10 cm x 10 cm, centred on the umbilicus, and a fenestrated drape placed.

Technique
- Abdominocentesis is performed using either a single centesis or a four-quadrant approach. In patients with smaller effusions, the chances of fluid retrieval can be increased using ultrasound-guided aspiration.
- The site for single abdominocentesis is a point approximately 1 cm lateral and to the right of the ventral midline and 1–2 cm caudal to the umbilicus (right cranial quadrant).
The needle should be inserted without a syringe attached. An open needle is more likely to result in fluid flow, as suction with a syringe will draw the omentum or viscera on to the needle.

1. Insert the needle through the skin and abdominal wall, and twist it slightly to encourage fluid flow.
2. Allow fluid to drip into a sampling tube.
3. If no fluid is obtained after 1 minute, apply slight negative pressure with a 5 ml syringe.
4. If fluid is still not obtained, repeat the procedure in three other sites: right caudal; left cranial (risk of splenic penetration); and left caudal.

5. If there is still a suspicion of abdominal fluid, diagnostic peritoneal lavage can be performed.

**Sample handling**

- Place fluid in an EDTA tube for total nucleated cell count and cytology.
- Place fluid in a plain tube for total protein and any other biochemical or serological tests.
- A sample in a sterile plain tube can be submitted for bacteriological culture if necessary.
- Make several fresh air-dried smears (unstained).

**Potential complications**

- Aspiration of blood: if blood is aspirated, stop the aspiration, place the blood in a glass tube and observe for clot formation. Blood from the abdominal cavity, i.e. haemorrhagic effusions, will not clot, whereas blood from a vessel or organ will clot. If bleeding persists, abdominal pressure should be applied by way of manual compression or a pressure bandage.

*Four sampling quadrants. X = needle position*
• Puncture of the GI tract: if fluid suggestive of GI contents is obtained, indicating that the GI tract has been punctured, any hole should seal when the needle is removed. The patient should, however, be monitored for developing peritonitis.
• Continued drainage after needle removal: in some animals with large abdominal effusions, the centesis hole may continue to drain fluid. If this occurs, a pressure dressing may be applied.

For interpretation of the results of fluid analysis, see the *BSAVA Manual of Canine and Feline Clinical Pathology*.

## ACTH response test

### Indications/Use

- Distinguishing spontaneous from iatrogenic hyperadrenocorticism
- Aid to diagnosis of hypoadrenocorticism (reliability identifies >50% of dogs with adrenal-dependent HAC and about 85% of dogs with pituitary-dependent HAC)
- Monitoring trilostane or mitotane therapy
- To aid the diagnosis of feline hyperadrenocorticism, especially when combined with the dexamethasone suppression test – (b) high dose

### Equipment

- Clippers
- Cotton wool or soft swabs
- 4% chlorhexidine gluconate or 10% povidone–iodine
- 70% surgical spirit
- Synthetic ACTH (0.25 mg/ml solution) (Tetracosactide, Tetracosactrin)
- Hypodermic needles: 21 G; ¾ to 1 inch
- Intravenous catheter
- 2 ml syringes
- Heparin and/or plain tubes

### Patient preparation and positioning

- The patient should be conscious.
- The patient is restrained in sternal recumbency or in a sitting position for collection of blood from the jugular vein and then for intravenous injection into the cephalic vein (see *Blood sampling – (b) venous*).
- The area over the vessel to be sampled is clipped.
- Using cotton wool or soft swabs, the clipped area is wiped with 4% chlorhexidine gluconate or 10% povidone–iodine followed by 70% surgical spirit.
Technique

1. Collect a blood sample (approximately 2 ml) from the jugular vein (see Blood sampling – (b) venous) and place into a heparin or plain tube to enable measurement of the basal cortisol concentration.

2. Inject 0.25 mg of synthetic ACTH into the cephalic vein. NB In dogs <5 kg and cats, use only 0.125 mg.

When dealing with a very small dose of synthetic ACTH, it is preferable to place an intravenous catheter to ensure that the entire dose is administered.

3. After 30–60 minutes, collect a second blood sample (approximately 2 ml) from the jugular vein and place into a separate heparin or plain tube.

4. Ensure the tubes are labelled correctly.

5. Separate the serum or plasma prior to sending the samples to the laboratory.

For interpretation of ACTH response test results, see the BSAVA Manual of Canine and Feline Endocrinology.

Anaphylaxis – emergency treatment

Identification

Anaphylaxis is an acute severe allergic reaction characterized by venous and arteriolar dilation and increased capillary permeability, which result in decreased venous return to the heart, hypotension and hypovolaemia. Signs of hypovolaemic shock may be associated with:

- Angioedema: this commonly results in facial swelling and swelling of the distal limbs but can include pharyngeal and laryngeal swelling
- Bronchospasm
- Pruritus
- Urticaria: raised red skin wheals or hives
- Vomiting.
Procedure
1. Establish and maintain an airway: intubate if necessary.
2. Check the animal’s breathing: administer 100% oxygen via a non-
   rebreathing mask if dyspnoea is present without airway obstruction.
3. Place a large intravenous catheter.
4. Adrenaline (0.02 mg/kg slowly i.v or into the trachea via an
   endotracheal tube if intravenous access is not available) should
   be given in life-threatening cases. Continuous monitoring of
   cardiovascular status for adrenaline-induced arrhythmias and
   hypertension and response to therapy is required.
5. Treat hypovolaemic shock with intravenous fluid therapy.
   Intravenous fluid therapy should be tapered to the individual and
determined by continuous cardiovascular and respiratory
   assessment of the patient to achieve and maintain cardiovascular
   stability. As a guide, shock boluses of crystalloids (90 ml/kg/h for a
   dog and 60 ml/kg/h for a cat) may be required initially.
   Supplemental boluses of colloids (10 ml/kg/h for a dog and
   6 ml/kg/h for a cat) may also be required during initial stabilization.
   Maintenance therapy may require a combination of crystalloids
   and colloids due to ongoing fluid losses into the interstitium.
6. In animals with hypotension confirmed by blood pressure
   measurement, which has not responded to steps 4 and 5,
   vasopressors such as dobutamine (5–15 µg/kg/min) or dopamine
   (3–10 µg/kg/min) may be required. Treatment with these drugs
   requires frequent or continuous blood pressure measurement.
7. In animals with bronchospasm and life-threatening angioedema,
   including laryngeal oedema, dexamethasone (1–2 mg/kg i.v.) and
diphenhydramine (0.5–1 mg/kg slow i.v. or i.m.) may be useful
   adjunctive treatments.
8. Identification and avoidance of the causative factor is important for
   long-term management.

For further discussion of the treatment of hypovolaemic shock see the BSAVA Manual of
Canine and Feline Emergency and Critical Care.

Arthrocentesis
Indications/Use
• Joint disease of unknown aetiology
• Pain on manipulation of a joint
• Joint effusion
• Joint heat
• Periarticular thickening
• Suspected immune-mediated joint disease (e.g. pyrexia of
  unknown origin)
• Suspected infective arthritis
• Monitoring response to therapy in infective arthritis and immune-mediated polyarthritis

**Equipment**
• As required for Aseptic preparation – (a) non-surgical procedures

Sterile gloves should be worn if the clinician wishes to palpate bony landmarks and the needle insertion site. As experience is gained, such palpation is not necessary and gloves may not be required, provided the needle insertion point is not touched.

• Hypodermic needles: 21–23 G; \( \frac{5}{8} \) to 1.5 inch, depending on joint size
• 2 ml syringes
• Microscope slides
• EDTA and heparin collection tubes
• Blood culture bottle/bacteriology swab in transport medium

**Patient preparation and positioning**
• Sedation or general anaesthesia are required.
• The patient is placed in lateral recumbency, with the affected joint uppermost.
• The joint for aspiration will often be dictated by the findings on clinical examination, e.g., pain on joint manipulation.
• Aseptic preparation – (a) non-surgical procedures is performed on an area approximately 5 cm x 5 cm over the site for arthrocentesis.

**Technique**
1. a. **Carpus**: The antebrachiocarpal joint is generally the most accessible. With the carpus fully flexed, the antebrachiocarpal joint is palpable as a depression just distal to the radius. Insert the needle medial to the common digital extensor tendon and cephalic vein, which pass over the centre of the joint space. The needle insertion site is just lateral to the tendon of extensor carpi radialis.
b. **Tarsus (hock):** The talocrural joint space is most readily aspirated. Flex and extend the talocrural joint to locate the position of the joint. With the joint in a neutral position, insert the needle on the dorsolateral aspect of the talocrural joint, just medial to the palpable lateral malleolus of the fibula, and advance it towards the plantaromedial aspect of the joint.

*Alternative:* The plantarolateral joint space may be aspirated by inserting the needle parallel to the calcaneus, just medial to the lateral malleolus.

c. **Stifle:** The joint capsule of the stifle has three sacs, all of which communicate. With the joint partially flexed, apply digital pressure to the joint capsule on the medial side of the patellar ligament. Introduce the needle lateral to the straight patellar ligament, midway between the femur and tibia. Direct the needle into the joint space, through the fat pad, towards the intercondylar space. If no synovial fluid is aspirated, the needle should be moved inwards or outwards and re-aspiration attempted, as the needle tip may be located in the fat pad.

*Alternative:* The needle can be inserted parallel to the straight patellar ligament, midway between the tibial tuberosity and patella. The tip of the needle is directed lateral to the patella in the lateral parapatellar joint pouch.

d. **Elbow:** The caudolateral approach is the easiest and most atraumatic. Flex the elbow to approximately 45 degrees, and palpate the lateral condyle of the humerus and the olecranon. Advance the needle parallel to the long axis of the ulna, midway between these two landmarks. The needle should slide between the lateral epicondylar crest of the humerus and the anconeal process of the ulna.
e. **Shoulder:** With the shoulder in a neutral position, palpate the acromion process and introduce the needle perpendicular to the skin directly below the acromion. Distal traction can be applied to the limb by an assistant to open the joint space if required. If the needle hits bone, gently ‘walk’ the needle a few millimetres proximally or distally until the joint is penetrated.

2. Attach a syringe to the needle and gently aspirate until synovial fluid appears in the hub of the needle.

3. Once sufficient fluid has entered the syringe (which may only be a hubful), release the suction to minimize inadvertent aspiration of blood, before withdrawing the needle and syringe. To reduce the chance of blood contamination further, the syringe can be removed from the needle prior to needle removal from the joint space.

**Sample handling**

- To maximize preservation of cell morphology, smears should be made immediately and allowed to air dry.
- If more than approximately 0.2–0.3 ml of fluid is obtained, this can be placed in an EDTA tube for cytological evaluation and total nucleated cell count.
- Heparin is the preferred anticoagulant for the mucin precipitation test and for measurement of viscosity.
- If bacterial infective arthritis is suspected, synovial fluid should be placed in blood culture media. Alternatively, a few drops of fluid can be placed on a sterile bacteriology swab placed in transport medium.

**Potential complications**

- Iatrogenic articular cartilage damage
- Joint infection

Details of the evaluation of synovial fluid are given in the *BSAVA Manual of Canine and Feline Clinical Pathology* and in the *BSAVA Manual of Canine and Feline Musculoskeletal Disorders*. 
Aseptic preparation – (a) non-surgical procedures

Indications/Use

- Skin preparation for any veterinary procedure requiring aseptic technique, where there is a risk of iatrogenic contamination leading to infection

Equipment

- Regularly maintained clean, sharp electric clippers
- Vacuum cleaner
- Cotton wool or soft swabs
- Container to hold used cotton wool or soft swabs
- Tap water
- Appropriate antiseptic
  - For routine preparation of healthy skin, 4% chlorhexidine gluconate or 10% povidone–iodine, used in combination with 70% surgical spirit, is appropriate
  - Chlorhexidine gluconate or povidone–iodine should be avoided in animals with known skin sensitivities to either of these antiseptics
  - Chlorhexidine gluconate is specifically toxic to the conjunctiva, cornea, meninges and middle and inner ear: 0.2% povidone–iodine solution without alcohol is recommended for preparation of the periocular area but must not be allowed to contact the cornea; 1% povidone–iodine solution without alcohol is recommended for preparation of the external ear canal for surgery
  - Alcohol has a desiccating effect when applied to mucous membranes and should not be used for surgeries close to, or involving, mucous membranes (e.g. intraoral surgery)
  - The application of antiseptics directly to open wounds is not recommended
- Appropriate sterile skin drapes; these may be:
  - well maintained and laundered cloth drapes
  - disposable semi-impermeable synthetic drapes
  - one sterile fenestrated drape may be appropriate for non-surgical procedures

Technique

1. Hair removal
   - Remove hair from the procedure site with electric clippers.
   - Include margins up to 15 cm around the proposed procedural site. Clip sufficient hair to avoid contamination of the veterinary surgeon, the procedural site and sterile equipment.
   - Remove clipped hair from the procedural site. A vacuum cleaner may be used with anaesthetized animals, but using a vacuum cleaner around a conscious or sedated animal may cause undue distress to the patient.
2. Skin preparation
   • Apply antiseptic of an appropriate concentration to balls of damp cotton wool or swabs.
   • Clean the clipped area by applying gentle pressure in a circular motion, beginning at the proposed procedural site (e.g. site of needle insertion) and working outwards in concentric circles. Use a fresh cotton wool ball or swab each time cleaning is returned to the centre of the procedural site.
   • Continue cleaning for at least 3 minutes until no dirt can be seen grossly on the cotton wool balls or swabs.
   • Dampen the hair around the clipped area to flatten it away from the procedural site.
   • Squirt or gently spray surgical spirit on to the procedural site.
   **NB** Do NOT use surgical spirit to prepare sites that include the eyes or mucous membranes.

3. Draping
   • Draping may not be required for short minor procedures where the risk of contamination of the procedural site and sterile equipment is negligible (e.g. fine needle aspiration of skin masses, arthrocentesis).
   • **Draping should always be used if in doubt.** A simple fenestrated drape laid over the site may be sufficient to prevent contamination of a procedural site. For short procedures and procedures performed in non-anaesthetized patients, towel clamps are, respectively, not required and not advised.

   Draping should be performed by a veterinary surgeon or nurse who has scrubbed hands and is wearing sterile gloves.

**Potential complications**
- Clipper rash
- Dermatitis due to idiosyncratic reaction to antiseptic
- Break in asepsis

**Aseptic preparation – (b) surgical procedures**

**Indications/Use**
- Skin preparation for all surgical procedures

**Equipment**
- Regularly maintained clean, sharp electric clippers
- Vacuum cleaner
- Cotton wool or soft swabs
• Container to hold used cotton wool or soft swabs
• Tap water
• Appropriate antiseptic
  – For routine preparation of healthy skin, 4% chlorhexidine gluconate or 10% povidone–iodine, used in combination with 70% surgical spirit, is appropriate
  – Chlorhexidine gluconate or povidone–iodine should be avoided in animals with known skin sensitivities to either of these antiseptics
  – Chlorhexidine gluconate is specifically toxic to the conjunctiva, cornea, meninges and middle and inner ear: 0.2% povidone–iodine solution without alcohol is recommended for preparation of the periocular area but must not be allowed to contact the cornea; 1% povidone–iodine solution without alcohol is recommended for preparation of the external ear canal for surgery
  – Alcohol has a desiccating effect when applied to mucous membranes and should not be used for surgeries close to, or involving, mucous membranes (e.g. intraoral surgery)
  – The application of antiseptics directly to open wounds is not recommended
• Appropriate sterile skin drapes: these may be well maintained and laundered cloth drapes, or disposable semi-impermeable synthetic drapes
  – 4 sterile field drapes are appropriate for most surgical operations
  – Sterile transparent adherent plastic drapes and sterile skin towels may be used at the veterinary surgeon’s discretion
• Sterile towel clamps

Technique
1. Hair removal
   • Remove hair from the surgical site with electric clippers.
   • Include margins of at least 15 cm around the proposed surgical incision.
   • Remove clipped hair immediately with a vacuum cleaner.
   • For operations on limbs, clip the entire limb apart from the phalanges. Phalanges should only be clipped when they are included in the surgical field. Wrap the unclipped phalanges and as much of the paw as the surgical site permits in a semi-impermeable, self-adhesive, non-adherent outer bandage material.

2. Skin preparation outside the operating room
   • Apply antiseptic of an appropriate concentration to balls of damp cotton wool or soft cotton swabs.
   • Clean the clipped area by applying gentle pressure in a circular motion, beginning at the proposed incision site and working outwards in concentric circles. Use a fresh cotton wool ball or swab each time cleaning is returned to the incision site.
• Continue cleaning until no dirt can be seen grossly on the cotton wool balls or swabs.
• For operations including the phalanges, soak the paw in a dilute solution of antiseptic for 3 minutes or until all gross dirt has been removed.
• Dampen the hair around the clipped area to flatten it away from the surgical site.

3. Patient transfer to the operating room
• Transfer the patient to the operating room.
• Position the patient appropriately for surgery, using sandbags and ties.
• For limb operations, the limb is hung from the toes or paw. This is carried out using ropes tied to vertical stands, with top horizontal crossbars that can be raised and lowered.

4. Skin preparation inside the operating room
• Apply antiseptic of an appropriate concentration to balls of damp cotton wool or swabs.
• Clean the clipped area by applying gentle pressure in a circular motion beginning at the proposed incision site and working outwards in concentric circles. Use a fresh cotton wool ball or swab each time cleaning is returned to the incision site.
• Continue this scrubbing for at least 3 minutes.
• Squirt or gently spray surgical spirit on to the surgical site. **NB Do NOT use surgical spirit to prepare surgical sites that include the eyes or mucous membranes.**

5. Draping
• Sterile surgical drapes are placed around the surgical field. Draping leaves exposed only aseptically prepared skin and covers all hair. Drapes are secured in place with towel clamps placed at each of the four corners of the surgical field and then along the sides of the surgical field as necessary.
• Sterile surgical skin towels may be attached to the edges of the skin incision using towel clamps following the initial skin incision. Such skin draping has not been shown to decrease the incidence of surgical site infection, but is preferred by some surgeons.
• A sterile transparent adherent plastic drape may be used to cover skin that is exposed following routine draping. This may decrease the incidence of surgical site infection and is preferred by many surgeons, especially for orthopaedic procedures.
• For limb operations, a non-scrubbed assistant passes the foot to the surgeon. The surgeon covers the foot with an appropriately sized sterile surgical drape, which is wrapped around the distal limb and secured with a towel clamp. The surgeon avoids contacting the foot directly, so as to remain sterile. The entire distal limb including the towel clamp and drape are wrapped in a semi-impermeable, self-adhesive,
non-adherent outer bandage material, which has been sterilized specifically for this purpose. The limb is then held by a sterile surgical assistant, while it is draped using four sterile surgical drapes as above.

Draping should be performed by a veterinary surgeon or nurse who has scrubbed their lower arms and hands and is wearing a mask, hat sterile gloves and gown.

**Potential complications**

- Clipper rash
- Dermatitis due to idiosyncratic reaction to antiseptic
- Break in asepsis
Barium contrast media

There is a wide variety of preparations of barium sulphate used in veterinary radiography. None of these preparations is authorized for veterinary use and most are POM.

<table>
<thead>
<tr>
<th>Formulation</th>
<th>Trade name</th>
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<td>1000 g pack</td>
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<td>900 g bottle</td>
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<td>Intropaste</td>
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<td>1200 g bottle</td>
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<tr>
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<td>HD 200 plus</td>
<td>312 g bottle</td>
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<td>105% w/v liquid barium suspension</td>
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<td>Liquid E-ZPaque</td>
<td>1900 ml</td>
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<td>210% w/v liquid barium suspension</td>
<td>Maxibar</td>
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<tr>
<td>98% w/v powder for suspension</td>
<td>E-Z-HD</td>
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<td>Barium impregnated polyethylene spheres</td>
<td>BIPS</td>
<td>Large (5 mm diameter) and small (1.5 mm diameter) combined in capsules</td>
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<tr>
<td>Barium sulphate powder</td>
<td>Barium sulphate BP</td>
<td>500 g</td>
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</table>
Use

Used for GI contrast studies. Barium sulphate has a higher atomic number than body tissues and therefore appears radiopaque. Barium sulphate is inert with no osmotic potential and is not absorbed or acted upon by alimentary secretions. Enables visualization of lumen of oesophagus and GI tract. Barium-impregnated polyethylene spheres (BIPS) may be used to assess intestinal transit times and gastric emptying rate and to detect obstructions. As with all contrast studies, plain radiographs should be taken prior to administration of barium. Barium paste is used for oesophageal studies as it adheres to mucosa. Barium can also be mixed with food to evaluate the oesophagus, and may demonstrate strictures or dilatations not seen with liquid barium or paste. Liquid barium may be used to evaluate any part of the GI tract and is used alone for evaluation of the stomach and intestines. Flavoured preparations designed for human use may be unpalatable for some animals. Mixing of barium with methylcellulose may improve the quality of GI tract studies.

Safety and handling

Skin irritation may occur with skin contact. Take care not to spill barium on the patient's coat; clean off any spills carefully.

Contraindications and adverse reactions

Barium is insoluble and should not be used outside the GI tract. If endoscopy is going to follow contrast radiography it is preferable to use a water-soluble contrast medium, as barium will prevent evaluation of the mucosa. Leakage of barium into body cavities may lead to granulomatous reactions or adhesions. If oesophageal or GI tract perforation is suspected, low-osmolar water-soluble contrast media should be used. Aspiration of barium during administration or as a result of vomiting or regurgitation can lead to aspiration pneumonia. May cause constipation, transient diarrhoea and abdominal pain.

Barium studies of the gastrointestinal tract – (a) oesophagus

Indications/Use

- Regurgitation
- Retching
- Dysphagia
- Vomiting of undigested food soon after eating

Particular care should be taken when administering barium to animals with swallowing disorders so as to minimize the risk of aspiration of contrast medium and subsequent inhalation pneumonia.
Contraindications
- Suspected or confirmed oesophageal perforation: use non-ionic iodinated contrast media instead

Equipment
- Barium sulphate (see Barium contrast media)
- Lukewarm water for dilution
- Soft tinned food
- Food bowl
- 20–60 ml syringe with catheter tip
- Radiographic equipment

Patient preparation and positioning
- The patient should be conscious if possible. Light sedation can be used if necessary, although this will increase the risk of contrast medium aspiration and may alter oesophageal function.
- Contrast medium should be administered orally, with the patient in a sitting or standing position.
- The animal is subsequently positioned for radiographs as detailed below.

Technique
1. Take plain lateral and ventrodorsal radiographs of the thorax and cranial abdomen. This is important, as significant findings such as foreign material may be masked by contrast medium.
2. Administer thick barium sulphate paste (60% w/v) orally, via a syringe at a dose of approximately 1 ml per 5 kg bodyweight.
   
   OR

   If a dilated oesophagus is suspected from the plain films, a more satisfactory technique is to encourage the patient to eat a mixture of soft tinned food to which has been added approximately 20–30 ml of 100% w/v barium sulphate suspension.

3. Immediately take lateral views (particularly left lateral) of the cervical and thoracic oesophagus. Occasionally, additional information may be gained from a dorsoventral or ventrodorsal view.
4. Re-administer the barium sulphate paste and repeat the radiographs if necessary.

Potential complications
- Aspiration pneumonia
- Vomiting
- Diarrhoea

Further information on contrast radiography of the oesophagus and its evaluation can be found in the BSAVA Manual of Canine and Feline Thoracic Imaging and the BSAVA Manual of Canine and Feline Gastroenterology.
Barium studies of the gastrointestinal tract – (b) stomach and small intestine

Indications/Use

- Persistent vomiting
- Haematemesis
- Displacement of the GI tract associated with diaphragmatic rupture
- Assessment of GI tract displacement by changes in size or position of adjacent organs
- Unexplained dilatation of the small intestine

Particular care should be taken when administering barium to animals with swallowing disorders so as to minimize the risk of aspiration of contrast medium and subsequent inhalation pneumonia.

Contraindications

- Suspected GI tract rupture
- Convincing evidence of small intestinal dilatation on plain films, with a strong suspicion of mechanical obstruction

Equipment

- Barium sulphate suspension (see Barium contrast media)
- Lukewarm water for dilution
- Soft tinned food
- Food bowl
- 20–60 ml syringe with catheter tip
- Radiographic equipment

Patient preparation and positioning

- For an elective examination, food should be withheld for 24 hours prior to the procedure.
- The patient should be conscious if possible. If sedation is necessary, acepromazine has been found to have the least effect on GI motility and will assist restraint and accurate positioning.
- Contrast medium should be administered orally, with the patient in a sitting or standing position.
- The animal is subsequently positioned for radiographs as detailed below.

Technique

1. Take plain lateral and ventrodorsal radiographs of the abdomen. This is important, as significant findings such as foreign material may be masked by contrast medium.
2. Administer approximately 10 ml/kg of barium sulphate suspension (30% w/v) orally via a syringe.

OR

Mix the barium suspension with food and encourage the patient to eat.

3. For a full evaluation of the stomach, take four views immediately following administration of the contrast medium: right lateral; left lateral; ventrodorsal; and dorsoventral. In each case the radiograph should be centred over the cranial abdomen at the level of the last rib.

4. To assess gastric emptying: take repeat radiographs of the stomach 5–10 minutes after the initial films to verify the presence of any observed lesions and to assess normal onset of gastric emptying. Further films at 10–15 minute intervals may be required in cases of delayed gastric emptying.

5. To evaluate the small intestine: following the gastric examination, take lateral and ventrodorsal views at regular intervals, depending on the rate of passage in the individual case, until either a diagnosis has been reached or the barium has entered the large intestine. A film taken 24 hours after administration of contrast medium should be used to demonstrate that all the contrast medium has reached the large intestine.

If the barium study is performed to demonstrate displacement of the GI tract during the investigation of an abdominal mass or diaphragmatic rupture, it is more economical to omit the initial films and take radiographs 30–45 minutes after administration of contrast medium. By this time the stomach and most of the small intestine should contain contrast medium.

Potential complications

- Aspiration pneumonia
- Vomiting
- Diarrhoea

Further information on contrast radiography of the stomach and small intestine and its evaluation can be found in the BSAVA Manual of Canine and Feline Abdominal Imaging and the BSAVA Manual of Canine and Feline Gastroenterology.
Barium studies of the gastrointestinal tract – (c) large intestine

Indications/Use
- Tenesmus
- Melaena
- Chronic diarrhoea
- Identification of the position of the large intestine in relation to caudal abdominal/intrapelvic masses

Contraindications
- Large rectal or colonic mass that would preclude the passage of the tube for administering barium sulphate
- Suspected rupture of the lower GI tract

Equipment
- Barium sulphate suspension (see Barium contrast media)
- Equipment for giving an enema and administering contrast medium: 20–60 ml syringe; lukewarm water or saline; enema pump or tubing and funnel; lubricating gel such as K-Y jelly
- Radiographic equipment
- Foley catheter
- Suture material

Patient preparation and positioning
- For an elective examination, food should be withheld for 24 hours prior to the procedure.
- A rectal examination is required to ensure that it is safe to insert the enema and barium sulphate.
- At least one non-irritant cleansing enema (e.g. warm water) should be given 2–3 hours prior to the procedure, to evacuate the large intestine. This can be performed with the patient in lateral recumbency or standing and is usually best performed outside.
- It may be preferable to sedate or anaesthetize the patient, and this will not interfere with the results of the contrast study.

Technique
1. Administer the diluted suspension of barium sulphate (20% w/v) slowly into the descending colon, using either an enema pump or a gravity-feed tube and funnel. The volume of contrast medium required is usually about 10 ml/kg bodyweight.
2. If the patient is sedated or anaesthetized, leakage of contrast medium from the anus may occur. This can be prevented by administering the contrast medium via a Foley catheter and securing the catheter within the rectum with a purse-string suture around the anus.
3. Take lateral and ventrodorsal radiographs of the abdomen. These do not have to be taken immediately, but generally within 30 minutes of barium administration.

4. Alternatively, a ‘double contrast’ study can be performed, which will allow much better evaluation of mucosal detail. This is achieved by distending the colon with air (approximately 5 ml/kg) after administration of barium (approximately 10 ml/kg of bodyweight) to coat the mucosa.

Potential complications
- Rupture of the lower GI tract
- Trauma to the lower GI tract
- Haematochezia
- Diarrhoea

Further information on contrast radiography of the large intestine and its evaluation can be found in the BSAVA Manual of Canine and Feline Abdominal Imaging and the BSAVA Manual of Canine and Feline Gastroenterology.

Biopsy see
- Bone biopsy
- Endoscopy of the gastrointestinal tract
- Rhinoscopy
- Skin biopsy
- Tissue biopsy – needle core

Blood coagulation tests see
- Buccal mucosal bleeding time
- Platelet count
- Whole blood clotting time

Blood film see
- Blood smear preparation

Blood gas analysis see
- Blood sampling – (a) arterial
Blood pressure measurement – (a) direct

Invasive blood pressure measurement by means of an arterial catheter is considered the ‘gold standard’ technique but is technically demanding in terms of placement of the catheter, maintaining patency of the arterial catheter and ensuring accurate ‘zeroing’ of the apparatus to ambient air at the level of the right atrium.

Indications/Use

- Monitoring arterial blood pressure in critically ill patients
- Monitoring arterial blood pressure during anaesthesia
- Arterial catheters can also be used for serial collection of arterial blood samples for blood gas analysis in animals with pulmonary disease

Contraindications

- Coagulopathy: arterial catheters may be placed but only with care and only into distal limb arteries
- Arterial catheters should not be placed at sites where risk of bacterial contamination and infection are high, e.g. due to local tissue damage, local skin infection, diarrhoea, urinary incontinence

Equipment

- No. 11 scalpel
- 20–22 G peripheral venous over-the-needle catheter
- T-connector or extension set containing heparinized saline (1 IU of heparin per ml of 0.9% saline)
- 70% surgical spirit
- Adhesive tape
- Soft padded bandage and outer protective bandage
- Non-compliant manometer tubing
- Pressure transducer: must be ‘zeroed’ to ambient air at the level of the right atrium
- Display monitor
- Pressurized continuous flush system

Patient preparation and positioning

- The patient should be positioned in lateral recumbency.
- The patient’s limb must be held still; this can be achieved by manual restraint.
- For monitoring of the anaesthetized patient, arterial catheters should be placed soon after anaesthetic induction, and before the animal’s blood pressure falls, as low BP makes palpation of a peripheral arterial pulse more challenging.
Sites

- The dorsal pedal artery in the hind paw is most commonly used.
- Other arteries that may be used include the femoral artery, auricular artery and palmar metacarpal artery in the forepaw.

Technique

1. Place a catheter into a peripheral artery.
   - Palpate the arterial pulse.
   - The skin overlying the artery is clipped, then sprayed or lightly wiped with surgical spirit. Excessive scrubbing/wiping of the skin should be avoided, as this may result in spasm of the artery.
   - Make a small stab incision in the skin overlying the arterial pulse.
   - A peripheral venous catheter is placed through the skin incision and then inserted into the artery using short, firm, purposeful movements to push the stylet and catheter through the muscular wall of the artery. For entry into the artery the catheter should be positioned at a slight angle to the artery – approximately 10–30 degrees.
   - The dorsal pedal artery runs at about 30 degrees to the long axis of the metatarsus from medial to lateral. During catheter placement, palpate the arterial pulse constantly proximal to the site of entry of the catheter into the artery. This allows the operator to guide the catheter tip towards the artery, which cannot be seen.
   - As soon as arterial blood is seen in the flash chamber of the catheter, the stylet and catheter are lowered to a position parallel to the artery and advanced together a little further into the artery, before the catheter is advanced over the stylet and completely into the artery.
   - Withdraw the catheter stylet and attach a T-connector or extension set containing heparinized saline to the catheter. Arterial blood should be seen to pulsate within the hub of the catheter or T-connector.
   - The catheter should be secured firmly in place with adhesive tape and covered with a bandage.

The bandage over arterial catheters must be labelled clearly to avoid inadvertent administration of fluids or drugs into an artery.
2. Connect the T-connector to a pressure transducer via non-compliant tubing filled with heparinized saline.

3. To allow trouble-free continuous monitoring (avoiding clotting in the arterial line), the set-up is combined with a pressurized continuous flush system. If this is not available, arterial catheters should be flushed hourly.

4. The transducer–monitor combination gives a continuous reading of blood pressure and shows the pressure waveform. Systolic and diastolic pressures are taken as the cyclic maximum and minimum pressures, respectively. Mean pressure is calculated automatically. Arterial blood pressure monitoring is usually continuous.

5. On removal of the catheter, apply direct pressure to the artery for 5 minutes, then cover with cotton wool or a gauze swab and adhesive tape.

**Potential complications**

- Excessive arterial bleeding/exsanguination following a failed attempt at catheterization or accidental removal of the catheter: firm pressure should be applied to the site for 10 to 15 minutes
- Vascular damage and subsequent tissue necrosis distal to the catheter. The risk of this complication can be minimized by:
  - Avoiding placing adhesive tape too tightly around the paw
  - Never using arterial catheters for giving drugs or fluids
– Regular monitoring of the catheter and paw: arterial catheters should be checked every hour
– Prompt removal of arterial catheters when they are no longer required

- Infection
- Thrombosis/thromboembolism
- Embolism of a piece of catheter due to accidental transection of the catheter with a blade or scissors
- Air embolism

Blood pressure measurement – (b) indirect

Non-invasive blood pressure measurements are technically less demanding than invasive measurements and can be rapidly applied in the emergency situation, although they might not fulfil the expectations of reliability and accuracy. There are two non-invasive methods in general use: the oscillometric method and the Doppler method. Both require a cuff.

Indications/Use
- To assess cardiovascular function
- Routine monitoring during anaesthesia

Equipment
- Doppler ultrasound probe
- Coupling gel
- Adhesive tape
- Inflatable cuff attached to a manometer
  OR
- Oscillometric blood pressure monitor with cuffs

The proper cuff width is 40% of the circumference of the site where the cuff will be placed. Cuffs that are too wide lead to falsely low readings; those that are too narrow lead to falsely high readings.

Patient preparation and positioning
- Can be performed on conscious, sedated or anaesthetized animals.

Assessment of general cardiovascular status should be made in the absence of sedative and anaesthetic drugs.
• For conscious animals, it is important that they are relaxed and that the limb used is not weight-bearing. Lateral recumbency, with the limb to be used uppermost, is often preferred.
• For the Doppler technique it is necessary to shave the appropriate site and apply adequate coupling gel.

**Technique**

**Doppler ultrasound**

An inflatable cuff attached to a manometer occludes an artery, and a piezoelectric crystal placed over the artery distal to the cuff detects flow. The re-entry of blood into the artery as the cuff is released causes a frequency change (Doppler shift) in sound waves, which is detected by the piezoelectric crystal and converted to a sound detected by the operator. *This method measures systolic pressure.*

1. Position the Doppler ultrasound probe over one of the following:
   • The palmar arterial arch, on the ventral aspect of the proximal metacarpal region
   • The plantar arterial arch, on the ventral aspect of the proximal metatarsal region
   • The median caudal artery on the ventral aspect of the tail.
2. Apply coupling gel directly to the transducer and position it so that the sound of flow is detected. Tape the transducer in place perpendicular to the artery.
3. Place the cuff around the limb proximal to the measurement site, avoiding the joints, or around the tail. The cuff should be applied snugly enough to allow insertion of only a small finger between the cuff and the leg or tail. Most cuffs have a mark that should be placed directly over the artery.

   *If the cuff is applied too tightly, the measurement will be erroneously low because the cuff partly occludes the artery; if applied too loosely, the measurement will be erroneously high because excessive cuff pressure will be required to occlude the artery.*

   The cuff must be prevented from moving down the leg or tail when inflated, either by flexing the carpus or tarsus, or by blocking distal movement of the cuff by placing a hand on the appendage, not on the cuff.
4. Infl ate the cuff to a pressure above the expected systolic pressure to occlude the artery. This will result in loss of the sound of flow.
5. Slowly deflate the cuff by a few mmHg per second until the sound of flow is again detected. At this time the cuff pressure is equal to the systolic pressure. In patients with very low systolic blood pressure (<70 mmHg), the value obtained may be closer to the mean rather than the systolic pressure.
Oscillometric technique
This uses a cuff to occlude the artery, and detects oscillations of the underlying artery when it is partly occluded. This system determines systolic, diastolic and mean arterial pressures. This method is less accurate in very small patients, patients with low blood pressure and patients with dysrhythmias. Muscle contractions also create oscillations and are a source of potential error.

1. Place the cuff snugly (see Step 3 above) over one of the following:
   - The radial artery proximal to the carpus
   - The saphenous artery proximal to the tarsus
   - The brachial artery proximal to the elbow
   - The median caudal artery at the base of the tail.

2. Attach the cuff to a control unit that continually senses arterial pressure and inflates to a pressure greater than systolic, and then automatically deflates the cuff.

3. The heart rate is displayed; verify that it matches the patient's heart rate by manually counting the heart rate by direct heart auscultation or palpation of an artery.

4. Record the values for 3–5 cycles and report the averages for systolic, diastolic and mean pressures.

Potential false readings
Incorrect blood pressure readings may be obtained due to:
   - Inappropriate cuff size
   - Inappropriate placement of the cuff
   - Excessive motion of the limb or tail
   - Low blood pressure
   - Dysrhythmias
   - Obesity
   - Peripheral oedema
   - Limb conformation, which does not permit snug placement of the cuff
   - Stress.

Blood sampling – (a) arterial

Indications/Use
   - To obtain a sample of arterial blood for assessment of respiratory function or acid–base status

Contraindications
   - Coagulopathy
   - Sampling should not be performed at sites where risk of bacterial contamination and infection are high, e.g. due to local tissue damage, local skin infection, diarrhoea, urinary incontinence
Equipment

- Hypodermic needle:
  - Cats: 23–25 G; 5/8 inch
  - Dogs: 23 G; 5/8 inch
- 2 ml syringe
- Heparin sodium: this is used to pre-coat the syringe and needle used for blood collection; approximately 0.5 ml of heparin sodium is aspirated into the 2 ml syringe via the needle and then expelled. Alternatively, pre-heparinized blood-gas syringes with needles attached can be used
- 70% surgical spirit
- Cotton wool or gauze swabs
- 25 mm wide adhesive tape

Patient preparation and positioning

- Arterial blood sampling is performed in the conscious animal.
- Sedation should be avoided if possible as it will affect the result of blood gas analysis.
- Animals should be positioned appropriately for the blood collection site (see below).

Sites

Most commonly the dorsal pedal artery is used, but in cats and small dogs it is sometimes easier to use the femoral artery.

Dorsal pedal artery

- The animal is placed in lateral recumbency, either on a table (cats and small dogs) or on the floor (large dogs), with the leg to be sampled placed closest to the table or floor.
- An assistant restrains the patient’s head with one hand and the uppermost hindlimb with the other.
- The artery is palpated just distal to the tarsus (hock), between the second and third metatarsal bones.
Femoral artery

- The animal is placed in lateral recumbency, either on a table (cats and small dogs) or on the floor (large dogs), with the leg to be sampled placed closest to the table or floor.
- The animal is restrained manually, and the upper limb abducted so that the femoral artery can be palpated.
- The femoral artery pulse is palpable on the medial thigh, ventral to the inguinal region and proximal to the stifle.

Technique

1. Stretch the skin over the artery.
2. Palpate the artery so that its pulsation can be felt.
3. The skin overlying the artery is clipped, then sprayed or lightly wiped with surgical spirit. Excessive scrubbing/wiping of the skin should be avoided, as this may result in spasm of the artery.
4. Direct the needle, with syringe attached, toward the palpated artery, at an angle of about 30 degrees. The needle bevel is pointed upwards.
5. Penetrate the artery in one quick firm purposeful movement.
6. When the artery has been penetrated, a flash of blood will be seen in the hub of the needle.
7. Collect approximately 1 ml of blood.
8. Remove the syringe and needle from the artery.
9. On removal of the needle, apply direct pressure to the artery for 5 minutes, then cover with cotton wool or a gauze swab and adhesive tape.
10. Hold the syringe upright and tap it to cause air bubbles to rise. Eject any air from the syringe.
11. Cap the sample with an airtight seal to prevent exposure to room air. Rubber bungs or plastic caps are available with pre-heparinized blood-gas syringes.
12. The blood sample must be analysed within 5 minutes.

Potential complications

- Significant haemorrhage is very uncommon, provided direct pressure is applied to the artery (see above)
- Bruising and the formation of a small haematoma will occur in some patients, but can be minimized by good technique and by the application of direct pressure to the artery
Arterial thrombosis is uncommon but is more likely if repeated attempts are made to collect blood from an artery.

Approximate normal arterial blood gas values for dogs and cats breathing room air are given below. See the *BSAVA Manual of Canine and Feline Clinical Pathology* for further details on interpretation of results.

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<td>7.31–7.46</td>
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<td>$PCO_2$</td>
<td>30.8–42.8 mmHg (4.10–5.69 kPa)</td>
<td>25.2–36.8 mmHg (3.35–4.89 kPa)</td>
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<tr>
<td>$PO_2$</td>
<td>80.9–103.3 mmHg (10.76–13.74 kPa)</td>
<td>95.4–118.2 mmHg (12.69–15.72 kPa)</td>
</tr>
<tr>
<td>$[HCO_3^-]$</td>
<td>18.8–25.6 mmol/l</td>
<td>14.4–21.6 mmol/l</td>
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<tr>
<td>Base excess</td>
<td>0 ± 4</td>
<td>0 ± 4</td>
</tr>
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Blood sampling – (b) venous

**Indications/Use**
- To obtain a sample of venous blood for clinical pathology tests or for bacterial culture

**Contraindications**
- Coagulopathy
- Sampling should not be performed at sites where risk of bacterial contamination and infection are high, e.g. due to local tissue damage, local skin infection, diarrhoea, urinary incontinence

**Equipment**
- Hypodermic needles:
  - Cats: 23–21 G; 5/8 inch
  - Dogs: 21 G; 5/8 in or 1 inch
- 2–10 ml syringes
- 70% surgical spirit
- 4% chlorhexidine gluconate or 10% povidone–iodine
- Cotton wool or gauze swabs
- 25 mm wide adhesive tape
- Appropriate blood containers (see table below) and/or three blood culture bottles (pre-warmed to 37°C)
- Sterile gloves
Patient preparation and positioning

- Venous blood sampling is performed in the conscious animal, although in fractious animals light sedation may be required.
- The animal should be positioned appropriately for the blood collection site (see below).
- Clip the hair over the appropriate vein.
- Using cotton wool or gauze swabs, clean the skin over the vein with 4% chlorhexidine or 10% povidone–iodine, followed by spraying with surgical spirit.
- When taking samples for bacterial culture, take care not to touch the site of needle insertion. If necessary, gloves should be worn.

Sites

The jugular vein is preferred to peripheral veins in order to minimize the potential for cell damage during blood sampling. The cephalic vein and the lateral saphenous vein (which runs over the lateral aspect of the hock) may sometimes be used.
**Jugular vein**
- The animal is placed in a sitting position, either on a table (cats and small dogs) or on the floor (large dogs).
- An assistant stands on the left of the patient.
- The assistant places their right arm over the patient’s back and round the front of the patient, to encircle and control the forelimbs.
- The assistant’s left arm is used to extend the animal’s neck by grasping its muzzle and directing the nostrils towards the ceiling.

**Cephalic vein**
- The animal is placed in a sitting position or in sternal recumbency, either on a table (cats and small dogs) or on the floor (large dogs).
- An assistant stands on the left of the patient.
- The assistant passes their left hand under the patient’s neck and holds the head turned away from the sampler.
- The assistant’s right arm is used to extend the patient’s right forelimb.

**Saphenous vein**
- The animal is placed in lateral recumbency, either on a table (cats and small dogs) or on the floor (large dogs).
- An assistant restrains the animal’s head with one hand.
- With the other hand, the assistant extends the uppermost hindlimb, at the same time stretching out the body. The hand is placed around the leg at the level of the mid-tibia/fibula.
Technique

For biochemical tests or haematology
1. Raise the vein by compressing it at a point closer to the heart than the venepuncture site.
2. Insert the needle, with syringe attached, into the vein with the bevel upwards, at an angle of approximately 30 degrees.
3. Aspirate blood by withdrawing the syringe plunger. Avoid excessive suction on the syringe as this may collapse the vein.
4. Release the pressure on the vein.
5. Remove the needle and apply gentle pressure to the venepuncture site for a few seconds.
6. If the cephalic or saphenous veins are used, apply a light bandage of cotton wool held by adhesive tape for 30–60 minutes.
7. Place the blood sample in the appropriate tube(s).
8. Gently invert the sample tube several times to ensure adequate distribution of any additive. Do NOT shake the tube, as this may cause haemolysis.

For bacterial culture
1. Follow steps 1 to 5 above to take a 5–10 ml blood sample (see culture bottle for required volume).
6. Place a new needle on the syringe.
7. Swab the rubber stopper of the culture bottle with surgical spirit and allow to dry.
8. Add the required volume of blood to the pre-warmed culture bottle.
9. Collect three blood samples with a minimum of 1 hour between samples OR, in acutely septic patients, all three samples can be taken over 30 minutes.
10. The culture bottles should be transported to the laboratory as quickly as possible. Although not ideal, overnight postage may still give meaningful results.

Potential complications
These are very uncommon but may include:
• Minor haemorrhage
• Subcutaneous haematoma formation
• Vascular trauma
• Thrombophlebitis

For information on interpretation of blood samples see the BSAVA Manual of Canine and Feline Clinical Pathology.
Blood smear preparation

Indications/Use

- Assessment of:
  - Leucocyte (WBC) differential count
  - Leucocyte abnormalities, e.g. toxic neutrophils, left shift, blast cells
  - Red blood cell morphology, e.g. polychromasia, anisocytosis, fragmented red cells, spherocytes, Heinz bodies, red cell parasites
  - Platelet count
  - Platelet abnormalities, e.g. macroplatelets and platelet clumps

Equipment

- Blood collected in an EDTA anticoagulant tube (see Blood sampling – (b) venous)
- Microhaematocrit tube
- Microscope slides
- A ‘spreader’ slide: this is narrower than the smear slide to avoid spreading the cells over the edge of the slide. ‘Spreaders’ can be made by breaking the corner off a normal slide, having first scored it with a blade or diamond writer

Technique

1. Using a microhaematocrit tube, place a drop of well mixed blood in the centre line toward one end of a microscope slide.
2. Hold the ‘spreader’ between the thumb and middle finger, placing the index finger on top of the ‘spreader’.
3. Place the ‘spreader’ in front of the blood spot, at an angle of about 30 degrees, and draw it backwards until it comes into contact with the blood, allowing the blood to spread out rapidly along the edge of the ‘spreader’.
4. The moment this occurs, advance the ‘spreader’ forwards smoothly and quickly.
5. As the smear is made, a ‘feathered edge’ forms. Do not lift the ‘spreader’ slide until the feathered edge is complete.
6. Ideally the smear should extend approximately two-thirds of the length of the slide.
7. Allow the smear to air dry fully before staining with an appropriate stain.

**Practical tips: common faults and how to avoid them**

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**Blood transfusion – (a) collection**

**Donor selection**

In the UK, the collection of blood from healthy animals is governed by the RCVS and Home Office guidelines. Readers are advised to consult the relevant documents prior to blood collection.

**Dogs**

- Healthy, fully vaccinated, not receiving medication (except flea preventive or routine worming medication)
- Suitable temperament
- >25 kg lean bodyweight
- 1–8 years of age
- Normal PCV, preferably >40%
- Ideally DEA 1.1 –ve
- Nulliparous
- No history of a previous blood transfusion
- No history of travel outside the UK
- Blood can be collected every 4–6 weeks without the need for iron supplementation
Cats
- Healthy, fully vaccinated, not receiving medication (except flea preventive or routine worming medication)
- Suitable temperament
- >4 kg lean bodyweight
- 1–8 years of age
- PCV >35%
- Blood typed (A, B or AB)
- No history of a previous blood transfusion
- No history of travel outside the UK
- Negative for FeLV, FIV, Mycoplasma felis
- Blood can be collected every 4–6 weeks without the need for iron supplementation

Equipment
- As required for Aseptic preparation – (a) non-surgical procedures
- Blood collection containers:
  - Dogs: standard commercial blood collection bag containing an anticoagulant, such as citrate phosphate dextrose (CPD), citrate phosphate dextrose adenine (CPDA) or acid citrate dextrose (ACD), attached to an extension tube and needle
  - Cats: blood collection bag specifically for feline patients containing an anticoagulant, such as CPD, CPDA or ACD, attached to an extension tube and needle OR two or three 20–30 ml syringes prefilled with anticoagulant (1 ml CPD, CPDA or ACD per 7 ml blood), and a 19 or 21 G butterfly catheter and extension tubing
- 500 ml bag of crystalloid fluids and intravenous catheter (for cats)
- Topical local anaesthetic cream (e.g. EMLA cream)
- Electronic scales (for weighing blood collection bags)
- Artery forceps
- Gauze swabs
- Clamping device and clamps
- Materials for a light neck bandage

Patient preparation and positioning
- Ensure that the donor meets the criteria listed above.
- Most dogs are able to donate blood without being sedated. Cats typically require sedation.

Examples of feline donor sedation protocols
- Butorphanol 0.1–0.2 mg/kg ± diazepam 0.5 mg/kg i.v.
- Ketamine 2 mg/kg and midazolam 0.1 mg/kg i.v.; additional boluses of one-quarter to one-half of the original dose as needed
- Ketamine 5–10 mg/kg and midazolam 0.2 mg/kg i.m. with additional boluses of ketamine 1.0 mg/kg i.v. as needed
- Mask isoflurane/oxygen anaesthesia
• Restrain dogs securely in lateral recumbency or a sitting position on a table.
• Restrain cats securely in sternal recumbency with the forelimbs over the edge of the table and head raised. Alternatively, restrain the cat in lateral recumbency with the neck outstretched.
• Apply topical local anaesthetic cream and wait approximately 20–30 minutes.
• Cats should have an **intravenous catheter** placed in a cephalic vein for the purpose of administering intravenous fluids following blood donation.

**Aseptic preparation – (a) non-surgical procedures** is carried out on the skin overlying the jugular groove.

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**Collection procedure**

**Dogs**

1. An assistant should apply pressure at the thoracic inlet to raise the jugular vein. Avoid contamination of the venepuncture site.
2. Remove the needle cap and perform venepuncture using the 16 G needle attached to the collection bag. If no flashback of blood is seen in the tubing, check needle placement and tubing for occlusion. The needle may need to be repositioned, but should not be fully withdrawn from the patient.
3. Position the bag lower than the donor to aid in gravitational flow and on a set of electronic scales.
4. Periodically invert the bag to ensure adequate mixing of blood and anticoagulant.
5. The maximum canine donation volume is approximately 16–18 ml/kg. The volume of blood that should be collected into a commercial blood bag is 450 ml, with an allowable 10% variance (405–495 ml). The weight of 1 ml of canine blood is approximately 1.053 g; therefore, the weight of an acceptable unit using one of these bags is approximately 426–521 g. When the bag is full, clamp the tubing with a pair of artery forceps and remove the needle from the jugular vein.
6. Using a gauze swab, apply pressure over the venepuncture site for 5 minutes. A light neck bandage should be applied for several hours.
7. Allow the tubing to refill with anticoagulant blood and clamp the distal (needle) end with a hand sealer clip or heat sealer. If these are not available, a knot can be tied in the line, although this is less desirable.
8. Clamp the entire length of tubing into 10 cm segments to be used for **cross-matching**.
9. Label the bag with the product type, donor identification, date of collection, date of expiration, donor blood type, donor PVC and phlebotomist identification prior to use or storage.
10. Following donation, food and water can be offered. Activity should be restricted to lead walks only for the next 24 hours, and it is advised that a harness or lead passed under the chest is used instead of a neck collar and lead, to avoid pressure on the jugular venepuncture site.
Cats

1. An assistant should apply pressure at the thoracic inlet to raise the jugular vein. Avoid contamination of the venepuncture site.
2. Remove the needle cap and perform venepuncture using the needle attached to the collection bag. If no flashback of blood is seen in the tubing, check needle placement and tubing for occlusion. The needle may need to be repositioned, but should not be fully withdrawn from the patient. Position the collecting bag lower than the donor to aid gravitational flow and on a set of electronic scales. Periodically invert the bag to ensure adequate mixing of blood and anticoagulant. Alternatively, use syringes containing anticoagulant and connected to a butterfly catheter. Without removing the butterfly catheter, perform venepuncture and fill each syringe in turn. The syringes can be rocked gently to ensure adequate mixing of blood and anticoagulant during collection. Invert the syringes several times after filling.
3. The maximum feline donation volume is approximately 11–13 ml/kg. The volume of blood that should be collected into a feline commercial blood bag is 50–60 ml. The weight of 1 ml of feline blood is approximately 1.053 g; therefore, the weight of an acceptable unit using one of these bags is approximately 52–63 g. When the bag is full, clamp the tubing with a pair of artery forceps and remove the needle from the jugular vein. Alternatively, if collecting blood into a syringe, remove the needle from the vein and cap the syringe when it contains the desired amount.
4. Using a gauze swab, apply pressure over the venepuncture site for 5 minutes. A light neck bandage should be applied for several hours.
5. If a blood collection bag is used, allow the tubing to refill with anticoagulant blood and clamp the distal (needle) end with a hand sealer clip or heat sealer. If these are not available, a knot can be tied in the line, although this is less desirable. Clamp the entire length of tubing into 10 cm segments to be used for cross-matching.
6. Label the bag or syringe with the product type, donor identification, date of collection, date of expiration, donor blood type, donor PVC and phlebotomist identification prior to use or storage.
7. Following donation, cats should receive intravenous fluid replacement in the form of 30 ml/kg of intravenous crystalloid solution over approximately 3 hours. The feline donor must be closely observed during recovery from sedation/general anaesthesia and may be offered food and water once fully awake.

Potential complications

- Haematoma
- Hypovolaemic shock
Storage

- Whole blood collected in a bag should be stored in a refrigerator maintained at 1–6°C with the bag in an upright position. Positioning the bag in this manner maximizes gas exchange with the red cell solution to help preserve the viability of the red blood cells during storage and following transfusion.
- Whole blood collected in a bag should be used within 28 days when collected in CPD or ACD, or within 35 days when collected in CPDA.
- Feline red blood cells collected in a syringe should be used within 5 hours.
- Whole blood can also be separated into packed red blood cells, fresh plasma, stored plasma and platelet-rich plasma concentrates. This should be done as soon as possible after collection, and plasma should be frozen within 8 hours to preserve coagulation and anticoagulation factors.

For details on separating whole blood and storage of the constituents, see the *BSAVA Manual of Canine and Feline Emergency and Critical Care* and the *BSAVA Manual of Canine and Feline Haematology and Transfusion Medicine*.

Blood transfusion – (b) cross-matching

Indications/Use

- To determine serological compatibility between a patient and donor blood

Dogs

Cross-matching should be performed whenever:

- The recipient has been previously transfused more than 4 days prior, even if a DEA 1.1-negative donor was used
- There has been a history of a transfusion reaction
- The recipient’s transfusion history is unknown
- The recipient has been previously pregnant

Cats

Cross-matching should be performed whenever:

- The recipient requires more than one transfusion, as previously transfused blood (even though it was the same AB type) may induce antibody production against red blood cell antigens separate from the AB blood group
- The donor or recipient blood type is unknown.
Equipment
• Approximately 5 ml of blood collected in EDTA anticoagulant from both the donor and recipient
• Centrifuge
• 5 ml plain plastic tubes
• 0.9% saline
• Pipette
• Microscope slides
• Microscope

Patient preparation and positioning: see Blood sampling – (b) venous

Technique
1. Collect blood from the jugular vein of the donor and recipient. Approximately 5 ml of blood from each should be placed into separate EDTA tubes (see Blood sampling – (b) venous). Alternatively, a sample of anticoagulated blood from the clamped donor blood tubing can be used.
2. Centrifuge the tubes (usually at 1000 RPM for 5–10 minutes), remove the supernatants (plasma) and transfer them to clean labelled 5 ml plain tubes (donor and recipient) for later use.
3. If a centrifuge is not available, allow the EDTA tubes to stand for ≥1 hour until the red blood cells have settled before using the supernatant.

Standard cross-match procedure
1. Wash the red blood cells three times with 0.9% saline and discard the supernatant after each wash.
2. Resuspend the washed red blood cells to create a 3–5% solution by adding 0.2 ml of red blood cells to 4.8 ml of saline (1 drop of red blood cells to 20 drops of saline).
3. For each donor prepare three tubes labelled as major, minor and recipient control.
4. Add to each tube 1 drop of the appropriate 3–5% red blood cells and 2 drops of plasma according to the following:
   a. Major cross-match = donor red blood cells and recipient plasma
   b. Minor cross-match = recipient red blood cells and donor plasma
   c. Recipient control = recipient red blood cells and recipient plasma.
5. Incubate the tubes for 15 minutes at room temperature.
6. Centrifuge the tubes at 1000 RPM for approximately 15 seconds to allow the cells to settle. Examine the samples for haemolysis (reddening of the supernatant).
7. Gently tap the tubes to resuspend the cells. Examine and score the tubes for agglutination.
8. If macroscopic agglutination is not observed, transfer a small amount of the tube contents to a labelled glass slide and examine for microscopic agglutination. This should not be confused with rouleaux formation.

9. For the recipient control:
   a. If there is no haemolysis or agglutination in the recipient control tube, the results are valid and incompatibilities can be interpreted.
   b. If there is haemolysis or agglutination present in the recipient control tube, then the compatibility and suitability of the donor cannot be accurately assessed.

**Rapid slide cross-match procedure**

An alternative and more rapid, but potentially less accurate, procedure for cross-match analysis involves visualizing the presence of agglutination on a slide rather than in a tube.

1. For each donor prepare three slides labelled as major, minor and recipient control.
2. Place 1 drop of red blood cells and 2 drops of plasma on to each slide according to the following:
   a. Major cross-match = donor red blood cells and recipient plasma
   b. Minor cross-match = recipient red blood cells and donor plasma
   c. Recipient control = recipient red blood cells and recipient plasma.
3. Gently rock the slides to mix the plasma and red blood cells. Examine for agglutination after 1–5 minutes.
4. For the recipient control: agglutination will invalidate results.

**Results of cross-matching**

- Any agglutination and/or haemolysis is a ‘positive’ result.
- A positive recipient control indicates that the patient is autoagglutinating. This makes interpretation of the test difficult, although it can be repeated with additional washing of the recipient’s red blood cells.
- A positive major cross-match indicates a significant antibody titre in the recipient against the donor red blood cells and precludes the use of that donor for transfusions.
- A positive minor cross-match indicates the presence of antibodies in the donor against the recipient red blood cells. If this reaction is strong, even small volumes of donor plasma may cause a significant transfusion reaction and precludes the use of the donor (unless red blood cells can be washed). With a weaker reaction, packed red blood cells from the donor may be transfused.
Blood transfusion – (c) typing

Indications/Use

- **Dogs:** As DEA 1.1 is the most antigenic blood type, it is strongly advised that the DEA 1.1 status of both the donor and recipient is determined prior to transfusion, or that only DEA 1.1-negative donors are used.
- **Cats:** *All donor and recipient cats must be blood typed* prior to transfusion, even in an emergency situation.

Equipment

- Approximately 2 ml of blood collected into an EDTA tube (see *Blood sampling – (b) venous*).
- Blood typing test kit.

Patient preparation and positioning: *see Blood sampling – (b) venous*.

Technique

Use a commercial blood typing test kit and follow manufacturer’s instructions.
Blood transfusion – (d) giving

Indications/Use

- Anaemia due to:
  - Haemorrhage
  - Haemolysis
  - Reduced erythropoiesis

Contraindications

- Administration of non-typed or non-cross-matched blood to a dog that has previously received a blood transfusion
- Administration of non-typed blood to a cat

Equipment

As required for Intravenous catheter placement

- Whole blood:
  - Dogs: As a general rule:
    - DEA 1.1-negative dogs should only receive DEA 1.1-negative blood
    - DEA 1.1-positive dogs may receive either DEA 1.1-negative or -positive blood
  - Cats:
    - Type A cats must only receive type A blood
    - Type B cats must only receive type B blood
    - The rarer type AB cats do not possess either alloantibody; they should ideally receive type AB blood, but when this is not available type A blood is the next best choice
• Dogs: Blood infusion set incorporating an in-line filter (170–260 µm) and suitable for connecting to a canine blood collection bag.
• Cats: Blood infusion set incorporating an in-line filter (170–260 µm) and suitable for connecting to a feline blood collection bag. Alternatively, blood collected in a syringe can be administered via an extension set. Again a filter should be used, such as a paediatric filter with reduced dead space or microaggregate filters of 18–40 µm.
• Intravenous catheter suitable for the size of the patient. A large-diameter catheter should be placed to avoid red cell haemolysis during blood administration.
• Adhesive tape

Patient preparation and positioning
• The patient should ideally be conscious, although sedation can be used if required. It is sometimes necessary to give blood to an anaesthetized patient intraoperatively.
• The patient should be placed on comfortable bedding and confined to a cage during the administration of blood.
• Patients should not receive food or medication during a transfusion, and the only fluid that may be administered through the same catheter is 0.9% saline.
• An intravenous catheter should be placed. Alternatively, blood can be given via an intraosseous needle if venous access cannot be obtained.

Technique
Blood is usually administered intravenously via an intravenous catheter, but it may also be given via the intraosseous route if venous access cannot be obtained (e.g. kittens, puppies). It should not be given intraperitoneally.

Volume
The amount of blood to be administered can be calculated as follows:
• As a ‘rule of thumb’:
  – 2 ml blood/kg bodyweight raises the PCV by 1%
• Suggested formulae for calculating the amount of whole blood required for transfusion are:
  – **Dog:**
    
    Volume of donor blood required = 
    
    recipient’s bodyweight (kg) x 85 x desired PCV – recipient’s PCV
    
    PCV of donated blood
– **Cat:**

Volume of donor blood required =

\[
\text{recipient’s bodyweight (kg) \times 60 \times \frac{\text{desired PCV} - \text{recipient’s PCV}}{\text{PCV of donated blood}}} \]

Total volume given should not exceed 22 ml/kg unless there are severe ongoing losses.

**Rate**

The rate of whole blood administration depends on the cardiovascular status of the recipient:

- In general, the rate should be only 0.25–1.0 ml/kg/h for the first 20–30 minutes
- If the transfusion is well tolerated, the rate may then be increased to deliver the remaining product within 4–6 hours
- In an animal with an increased risk of volume overload (cardiovascular disease, impaired renal function), the rate of administration should not exceed 3–4 ml/kg/h.

**Monitoring**

The following parameters should be measured prior to (‘baseline’), every 15–30 minutes during, and 1, 12 and 24 hours after transfusion:

- Demeanour
- Rectal temperature
- Pulse rate and quality
- Respiratory rate and character (see Cardiorespiratory examination)
- Mucous membrane colour and capillary refill time
- Plasma and urine colour.

- PCV and TP should also be monitored prior to, upon completion of, and at 12 and 24 hours after transfusion.

**Adverse reactions and action required**

**Acute haemolytic reaction with intravascular haemolysis**

- Seen in type B cats receiving type A blood as well as in DEA 1.1-ve dogs sensitized to DEA 1.1 upon repeated exposure.
- Clinical signs may include fever, tachycardia, dyspnoea, muscle tremors, vomiting, weakness, collapse, haemoglobinaemia and haemoglobinuria.
- May lead to shock, disseminated intravascular coagulation, renal damage and, potentially, death.
- **Treatment involves immediate discontinuation of the transfusion** and treatment of the clinical signs of shock.
Non-haemolytic immunological reactions
- Acute type I hypersensitivity reactions (allergic or anaphylactic), most often mediated by IgE and mast cells.
- Clinical signs including urticaria, pruritus, erythema, oedema, vomiting and dyspnoea secondary to pulmonary oedema.
- Treatment involves immediate discontinuation of the transfusion and evaluation of the patient for evidence of haemolysis and shock. Steroids (dexamethasone 0.5–1.0 mg/kg i.v.) and antihistamines (chlorphenamine 4–8 mg q8h for dogs; 2–4 mg q8–12h for cats) may be required.

Further information on blood transfusion can be found in the BSAVA Manual of Canine and Feline Haematology and Transfusion Medicine and the BSAVA Manual of Canine and Feline Emergency and Critical Care.

Bone biopsy – needle

Indications
- To obtain a sample of bone in suspected cases of:
  - Primary bone tumours
  - Metastatic bone lesions
  - Systemic mycosis with bony localization
  - Bacterial osteomyelitis
- To obtain a bone marrow core sample

Contraindications
- Fracture in the region to be sampled
- Coagulopathy

Equipment
- Jamshidi bone biopsy needle:
  - 12 G for dogs >5 kg
  - 14 G for dogs <5 kg and cats
- As required for Aseptic preparation – (a) non-surgical procedures
- No. 11 or 15 scalpel
- Suture materials or tissue adhesive for skin closure
- Bone biopsy collection tubes (with 10% buffered formalin)
- Plain sterile pots for culture
Patient preparation and positioning

- General anaesthesia is required.
- Position the animal with the area to be sampled uppermost.
- For bone biopsy, the centre of the lesion is chosen, identified via radiography. Biopsy at the lesion periphery will often result in sampling the reactive bone surrounding the primary lesion.
- **Aseptic preparation – (a) non-surgical procedures** is performed at the site on an area approximately 5 cm x 5 cm.

Technique

1. Make a small skin incision with a scalpel blade over the site of needle insertion.
2. With the stylet of the Jamshidi needle in place, advance the cannula through the soft tissues until the bone is reached.

   *Advance the needle to the bone*

3. Remove the stylet and penetrate the bone cortex with the cannula.
4. Advance the cannula a sufficient distance into the bone (1.5–3 cm).
5. Vigorously rotate the cannula in one direction and also swiftly move it sideways, to ensure the biopsy sample is sectioned at its base.
6. Remove the cannula.
7. Insert the blunt probe retrograde into the tip of the cannula to expel the specimen through the base.

   *Removing the sample*
8. Place the specimen into an appropriate collection pot.
9. Repeat the procedure with redirection of the instrument to obtain multiple core samples.
10. The skin incision should be sutured or closed with tissue adhesive.

Sample handling
- The sample should be fixed in 10% neutral buffered formalin.
- The sample should be sent to a histopathology laboratory. Note: the sample will have to be decalcified at the laboratory, a process that may take several days.

Potential complications
- It is possible that bacterial or fungal infections may spread into the surrounding soft tissue during bone biopsy
- Rarely, bone can be weakened enough for a fracture to occur. This is more likely to happen if multiple samples are taken

Bone marrow aspiration

Indications
To obtain a sample of bone marrow for cytology to aid diagnosis in:
- Non-regenerative anaemia
- Neutropenia or thrombocytopenia
- Unexplained leucocytosis, polycythaemia or thrombocytosis
- Excessive numbers of cells with abnormal morphology in the peripheral blood
- Pyrexia of unknown origin
- Hyperproteinaemia associated with a monoclonal or polyclonal gammopathy
- Unexplained hypercalcaemia
- Multicentric lymphoma

Contraindications
- Coagulopathy

Equipment
- As required for Aseptic preparation – (a) non-surgical procedures
- Klima or Rosenthal needle with interlocking stylet:
  - 14 G for most dogs
  - 16 G for dogs <5 kg and cats
- 20 ml syringe
Procedures in Small Animal Practice

- Local anaesthetic
- Scalpel
- Microscope slides
- EDTA collection tube
- Container with 10% buffered formalin
- Tissue glue or suture materials for skin closure

Patient preparation and positioning
- In dogs, sedation and local anaesthesia are usually sufficient, although the more inexperienced clinician may prefer general anaesthesia.
- In cats, general anaesthesia should be used.
- Aseptic preparation – (a) non-surgical procedures is performed on an area approximately 5 cm x 5 cm.
- Infiltrate 1–2 ml of local anaesthetic into the skin, subcutis and periosteum.

Technique
1. Make a small stab incision in the skin over the required site.
   - **Dogs:** Iliac crest; greater tubercle of humerus; or trochanteric fossa of the femur.
   - **Cats:** Greater tubercle of humerus; or trochanteric fossa of the femur.
2. Introduce a Klima or Rosenthal needle, with stylet in place, through the subcutis and on to the bone:

   **Iliac crest:** Place patient in sternal recumbency with both hind limbs pushed tightly under the body. Insert the needle into the most dorsal palpable aspect of the iliac crest.

Practical tip
To prevent the bone marrow clotting, the collection syringe may be pre-loaded with 0.3 ml of 3% EDTA solution or ACD (acid citrate dextrose removed from a blood collection bag).
**Femur:** With the patient in lateral recumbency, palpate the greater trochanter and insert the needle medial to this and into the trochanteric fossa. Once the needle is in the trochanteric fossa, advance it parallel to the shaft of the femur.

**Humerus:** With the patient in lateral recumbency, insert the needle on the craniolateral aspect of the greater tubercle of the humerus.
3. Advance the needle by rotating it to and fro in one plane, whilst applying firm pressure until it enters the medullary cavity. Entry into the medullary cavity is detected as a decrease in resistance to cannula insertion into the bone or increased stability of the cannula within the bone. When the cannula is properly seated within the medullary cavity, movement of the cannula will result in the same movement of the bone.

4. Remove the stylet and attach a 20 ml syringe to the needle.

5. Aspirate with several quite forceful withdrawals of the plunger. The animal may show a transient pain response as the marrow is aspirated.

6. Release the plunger as soon as marrow appears in the syringe.

7. Remove the syringe, leaving the needle in place.

8. Process the bone marrow sample immediately, as detailed below.

9. If no bone marrow is obtained this may be due to poor needle placement or to marrow fibrosis. Advance the needle a few millimetres and re-apply suction. If this is still not successful, withdraw the needle, replace the stylet and redirect the needle. When two or three attempts have been unsuccessful, an alternative site should be found.

10. Optional: After obtaining marrow, the needle may be left in place without the syringe. It can then be used to obtain a small core biopsy sample, by advancing it a further 10–20 mm into the marrow cavity, without the stylet in place. Rotate the needle vigorously in one direction and then remove it. The core sample is removed from the needle using a blunt probe or, if not available, the stylet.

11. Close the skin deficit with tissue glue or a single suture.

Sample handling

- Prior to aspiration, place several microscope slides at a near-vertical angle.

- Place a drop of marrow at the top end of the slides. Blood runs down to the bottom of the slide, whilst the marrow spicules should remain on the slide. Alternatively, the bone marrow can be squirted into a Petri dish containing an anticoagulant such as EDTA or ACD (acid citrate dextrose) removed from a blood collection bag. The spicules, which should float, can then be removed with forceps.

- Smears of the marrow spicules can be made using the squash preparation technique (see Fine needle aspiration). Smears should be made quickly as the marrow clots rapidly (usually within 10–20 seconds).

- Unstained smears should be submitted to a laboratory for cytological analysis.

- Excess marrow can be placed in an EDTA tube for cytological evaluation, although morphology will change with time.

- If a core biopsy sample is obtained, place this in 10% buffered formalin.
Potential complications
- Significant haemorrhage is unlikely. However, in a severely thrombocytopenic patient, prolonged digital pressure should be applied to the biopsy site
- Puncture or laceration of muscles and nerves
- Sciatic nerve damage: this can be avoided during placement of a femoral cannula by walking the needle off the medial edge of the greater trochanter

Details of the cytological evaluation of bone marrow are given in the BSAVA Manual of Canine and Feline Haematology and Transfusion Medicine and the BSAVA Manual of Canine and Feline Clinical Pathology.

Bone marrow biopsy see
- Bone marrow aspiration

Bronchoalveolar lavage

Indications
- To obtain a sample for cytology and bacteriology from the lower airways of dogs and cats, especially those with deep parenchymal disease

BAL is used to sample smaller airways (lower bronchioles and alveoli) and can be performed using an endoscope or ‘blind’. The upper airways of medium and large-sized dogs may also be sampled by transtracheal wash. The upper airways of dogs and cats may also be sampled by endotracheal wash.

Contraindications
- As for Bronchoscopy

Equipment
- As for Bronchoscopy
- Aspiration/lavage catheter
- 500 ml warm 0.9% sterile saline
- 5–20 ml syringes
- Hypodermic needles: 21 G
- Microscope slides
- EDTA and sterile plain collection tubes
Patient preparation and positioning
As for Bronchoscopy.

Technique
1. Perform bronchoscopy.
2. Once the lung lobes to be sampled have been selected, pass the bronchoscope into successively smaller airways until it sits snugly.
3. Pre-draw sterile saline into several syringes.
4. Instil sterile saline via the endoscope channel:
   - One bolus of 20 ml is used in dogs >10 kg
   - One bolus of 10 ml is used in dogs <10 kg and in cats.
5. Gently suck the saline back, using the same channel, into a syringe.
6. Negative pressure during aspiration indicates the need to decrease suction to avoid airway collapse. If necessary the bronchoscope can be repositioned slightly, taking care not to dislodge the tip of the bronchoscope from the airway in which it is wedged.
7. Repeat steps 4 to 6 as necessary.
8. Ideally, at least two lung lobes should be sampled.
9. Alternatively, the procedure can be performed using a lavage catheter passed through the biopsy channel of the endoscope or adjacent to the endoscope.

Sample handling
- Ideally, 40–90% of the fluid instilled should be retrieved.
- Recovered fluid is typically slightly turbid, with a foamy layer at the top, representative of surfactant.
- Submit a portion of the sample in a sterile plain tube for culture.
- Place an aliquot in an EDTA tube for cytology.
- Fresh air-dried smears of any flocculent/mucoid material can also be made and submitted to the laboratory for staining (see Fine needle aspiration).

Potential complications
- Larynx or airway spasm
- Catheter breakage and aspiration of the catheter into the airway
- Worsening of respiratory status due to stress

Performing coupage during the procedure may assist sample retrieval.

Further information on endoscopic equipment and techniques can be found in the BSAVA Manual of Canine and Feline Endoscopy and Endosurgery. Further information on bronchoscopy and its interpretation can be found in the BSAVA Manual of Canine and Feline Cardiorespiratory Medicine.
Bronchoscopy

Indications
- Evaluation of tracheal and lower airway disorders
- Acquisition of samples (see also Bronchoalveolar lavage)
- Foreign body removal

Contraindications
- Severe hypoxaemia
- Coagulopathy
- Severe cardiac arrhythmia/dysfunction
- Partial tracheal obstruction
- Unstable asthma
- Pulmonary hypertension

Equipment
- Flexible endoscope: diameter 2.5–5 mm, length 25–80 cm
- Endoscopic viewing equipment
- Topical anaesthetic
- Mouth gag
- Swivel T-adaptor
- Supplementary oxygen
- Foreign body retrieval forceps including basket, rat-toothed, alligator, net and polyp snare-type

Patient preparation and positioning
- Inhalation anaesthesia is generally recommended, but if the patient is too small to have an endoscope passed through an endotracheal (ET) tube, intravenous anaesthesia must be used. Intubation should only be performed if the endoscope can fit easily through the ET tube, allowing for movement of air and the endoscope at the same time. A swivel T-adaptor can be used to provide constant gas anaesthesia whilst the endoscope is passed through the ET tube.
- Pre-oxygenation is very helpful, especially when there is compromised oxygenation. This can be provided through nasal oxygen delivery or a face mask. Supplementary oxygen can also be delivered through the channel of the endoscope or via a catheter placed alongside the endoscope. Flow volumes of 1–3 litres per minute can be safely used.
- Position the patient in sternal recumbency with the head elevated and neck extended.
- A mouth gag is essential to keep the mouth open and prevent the patient biting down on to the endoscope in the event of contact with the pharynx stimulating the gag reflex.
• Endoscopes should not remain in an airway for longer than 30–50 seconds, as they can interfere with ventilation and could result in hypercapnia and overventilation of the lungs, trauma and bronchospasm.

• If oxygen is being delivered through the channel constantly, carbon dioxide can not escape through the same channel.

Technique
1. Spray the larynx with topical anaesthetic to avoid laryngospasm.
2. Advance the endoscope into the larynx and examine this region.
3. If the patient is to be intubated, the proximal trachea should be evaluated prior to intubation. Intubation can be performed after evaluation of the length of the trachea that would otherwise be covered by the ET tube.
4. Centre the endoscope as it is advanced, and take care not to irritate the surface of the trachea with the endoscope.
5. As the endoscope is advanced, the carina or bifurcation is seen. The patient’s right side is on the operator’s left side; therefore the right mainstem bronchus will be seen on the left side of the image. The left and right mainstem bronchi branch off crisply with sharp edges.
6. The right mainstem bronchus is in line with the trachea and should be examined first.
7. Then pass the endoscope into the left mainstem bronchus.
8. Evaluate segmental and subsegmental airways on the left and right sides as thoroughly and systematically as possible.
9. Following bronchoscopy, the patient should remain intubated and allowed to breathe 100% oxygen for 10 minutes. Pulse oximetry should be utilized to measure the patient’s oxygenation throughout the recovery from anaesthesia and during the postoperative procedure.

Extra care should be taken when performing bronchoscopy in cats, as their airways are particularly prone to bronchospasm. The procedure should be performed as quickly as possible, with the minimum of trauma. Supplementary oxygen is highly recommended both before and after the procedure, and administration of a bronchodilator should be considered.

Foreign body removal
1. Position the endoscope several centimetres proximal to the foreign body.
2. Pass the retrieval forceps, in the closed position, down the biopsy channel (if there is sufficient room) or adjacent to the endoscope.
3. Open the forceps, grasp the foreign body and close the forceps.
4. Remove the endoscope and forceps at the same time.

- Care must be taken to provide adequate ventilation during the retrieval procedure.
- Foreign body removal can be very challenging. The veterinary surgeon should be prepared to stop and refer the animal for surgery if the procedure becomes prolonged.

Potential complications
- Bronchospasm: the chance of this occurring can be reduced by pre-treatment with terbutaline (0.01 mg/kg s.c. 30 mins prior to the procedure)
- Laryngospasm and coughing
- Hypoxaemia
- Haemorrhage (uncommon)
- Infection (rare)
- Pneumothorax

Further information on endoscopic equipment and techniques can be found in the BSAVA Manual of Canine and Feline Endoscopy and Endosurgery. Further information on bronchoscopy and its interpretation can be found in the BSAVA Manual of Canine and Feline Cardiorespiratory Medicine.

Buccal mucosal bleeding time

Indications
- Suspected primary coagulopathy

Contraindications
- Thrombocytopenia
- Known primary haemostatic abnormality

Equipment
- Spring-loaded bleeding device (e.g. Simplate)
- 2 cm wide gauze bandage
- Tissue paper/filter paper
- Stopwatch or timer
Patient preparation and positioning

- If possible, the procedure should be performed with the animal conscious.
- Light sedation may be required in fractious dogs and in cats.
- The patient is restrained in lateral or sternal recumbency.

Technique

1. Fold back the patient's upper lip and hold it in place, either via an assistant or with a gauze bandage, causing moderate engorgement of the mucosal surface.
2. Position the bleeding device on the buccal mucosa, avoiding any obvious superficial vessels. Hold firmly but avoid excessive pressure.
3. Depress the trigger on the device to create a small incision in the buccal mucosa and simultaneously start the timer. Remove the bleeding device approximately 1 second after triggering.
4. At 15 seconds, blot the flow of blood with filter paper placed 1–3 mm below the incision without dislodging the clot.
5. Blot in a similar manner every 15 seconds until blood no longer stains the filter paper.
6. Stop the timer when bleeding has ceased.
7. Record the time from making the incision to cessation of bleeding.

Results

- In healthy dogs, BMBT is 1.7–3.3 minutes; this can be mildly prolonged (to 4.2 minutes) in anaesthetized or sedated dogs.
- BMBT of healthy anaesthetized cats is <3.3 minutes.
- Prolonged BMBT may indicate thrombocytopenia (<75 x 10⁹/l), thrombopathias (e.g. aspirin-induced), or von Willebrand's disease.

Potential complications

Prolonged bleeding can occur (uncommon) but should cease with continued pressure over the incision site. If it does not, the administration of fresh frozen plasma may be required.

Further details on coagulation tests and abnormalities can be found in the BSAVA Manual of Canine and Feline Haematology and Transfusion Medicine.
Cardiopulmonary–cerebral resuscitation

When to resuscitate

- The decision to begin CPCR is based upon clinical signs, consideration of the potential outcome, and a previous agreement (if possible) with the owner
- Resuscitation should only be attempted in patients that have a treatable disease
- CPCR is not advisable in animals in the terminal stages of an incurable disease (hepatic, renal, cardiac failure, etc.) or in those that have suffered severe head trauma with brain damage, where there is no reasonable chance of restoring near-normal mentation. In these circumstances, the owner’s permission not to resuscitate should be sought at the first presentation and the whole veterinary team made aware of their wishes (e.g. by using an appropriate notice on the kennel)

Equipment

- Pressure bag for rapid fluid infusion
- 50% dextrose
- Lactated Ringer’s solution
- Ambu bag
- Endotracheal (ET) tubes, various sizes
- Laryngoscope
- Hypodermic needles, various sizes
- Assorted intravenous catheters
- Gauze sponges/swabs
- 25 mm wide adhesive tape
- 50 mm roll of gauze
- Polyethylene urinary catheters
- Suture materials
- 3-way taps
- Thoracostomy tray
- Clippers
- 4% chlorhexidine gluconate or 10% povidone–iodine
- 70% surgical spirit
- Electrocardiogram monitor, leads, clips and conduction gel
- Doppler blood pressure monitor
- Defibrillator
- Drugs (see table overleaf)
<table>
<thead>
<tr>
<th>Drug</th>
<th>Indications</th>
<th>Dosage</th>
</tr>
</thead>
<tbody>
<tr>
<td>Adrenaline</td>
<td>Severe bradycardia, Ventricular fibrillation, Ventricular asystole, Pulseless electrical activity</td>
<td>0.01–0.2 mg/kg i.v. bolus q3–5min, 0.04–0.4 mg/kg intratracheal, 0.1–1 µg/kg/min i.v. CRI</td>
</tr>
<tr>
<td>Atropine sulphate</td>
<td>Sinus bradycardia, Atrioventricular block, Ventricular asystole</td>
<td>0.04 mg/kg i.v, 0.4 mg/kg intratracheal</td>
</tr>
<tr>
<td>Calcium gluconate (10%)</td>
<td>Hyperkalaemia, Hypocalcaemia, Calcium channel-blocker toxicity, Hypermagnesaemia</td>
<td>0.5–1.0 ml/kg i.v. to effect; closely observe the ECG</td>
</tr>
<tr>
<td>Diltiazem</td>
<td>Supraventricular tachycardia, Ventricular fibrillation, Hypertrophic cardiomyopathy</td>
<td>Dogs, cats: 0.25 mg/kg i.v. bolus, to cumulative dose of 0.75 mg/kg</td>
</tr>
<tr>
<td>Dobutamine</td>
<td>Myocardial failure, Low cardiac output</td>
<td>5–20 µg/kg/min CRI</td>
</tr>
<tr>
<td>Dopamine</td>
<td>Bradycardia, Low cardiac output, Hypotension</td>
<td>5–10 µg/kg/min CRI for increased contractility and cardiac output, 10–20 µg/kg/min CRI for vasoconstriction</td>
</tr>
<tr>
<td>Furosemide</td>
<td>Cerebral/pulmonary oedema, Congestive heart failure, Hypertension, Oliguria/anuria</td>
<td>Dogs: 2–4 mg/kg i.v., i.m., Cats: 1–2 mg/kg i.v., i.m.</td>
</tr>
<tr>
<td>Lidocaine</td>
<td>Ventricular tachycardia, Ventricular fibrillation</td>
<td>Dogs: 2–8 mg/kg i.v. bolus followed by 30–80 µg/kg/min CRI, Cats: 0.25–0.5 mg/kg i.v. bolus followed by 10–20 µg/kg/min CRI</td>
</tr>
<tr>
<td>Mannitol</td>
<td>Cerebral oedema, Oliguria</td>
<td>0.5–1.0 mg/kg i.v. given slowly over 10 min</td>
</tr>
<tr>
<td>Morphine sulphate</td>
<td>Analgesia/sedation, Vasodilator, Pulmonary oedema</td>
<td>0.04–0.08 mg/kg i.v., i.m., s.c.</td>
</tr>
<tr>
<td>Naloxone</td>
<td>Electromechanical dissociation, Narcotic overdose</td>
<td>0.03 mg/kg i.v. or intrathecal</td>
</tr>
<tr>
<td>Sodium bicarbonate</td>
<td>Severe metabolic acidosis</td>
<td>0.5–1.0 mEq/kg i.v.</td>
</tr>
</tbody>
</table>

**Technique**

**Phases of basic life support**

Once the decision to perform CPCR has been made, basic and advanced life support should be initiated as rapidly as possible in a sequential, orderly and predetermined manner.
Basic life support includes:
A  establishing and maintaining an Airway
B  controlling Breathing
C  Circulatory support via the initiation of manual chest compression
D  Drugs
**Airway**

- Check the airway for mucus, soft tissue structures (soft palate, tongue) or foreign material, such as food, causing an obstruction.
- Perform endotracheal intubation with a cuffed ET tube. The head should be extended and pulled forward prior to endotracheal intubation.
- When airway obstruction is severe or cannot be removed, **tracheostomy** should be performed.
- In animals with incomplete upper airway obstruction, transtracheal catheter ventilation is less traumatic. Ventilation is achieved by transcutaneously puncturing the trachea with a catheter. The catheter is attached to venous extension tubing and oxygen provided at flow rates of approximately 50 ml/kg.
- **Tracheostomy** generally remains the technique of choice, because it may allow spontaneous breathing of room air until the obstruction can be resolved.

Placement of a tracheostomy tube in a dyspnoeic patient with upper airway obstruction in an emergency situation by an *inexperienced* veterinary surgeon may not result in optimal results. Stabilization with oxygen therapy, sedatives (e.g. butorphanol plus acepromazine) and/or emergency anaesthesia and endotracheal intubation, prior to tracheostomy tube placement, may give a better outcome.

**Breathing**

- Controlled or assisted ventilation can be accomplished by connecting a properly placed ET tube to a self-inflating Ambu bag, demand valve or anaesthetic machine.
- Respiratory rate should be between 6 and 12 breaths per minute in animals with normal lungs.
- Ratios of 1 breath to 5 chest compressions or 2 breaths per 10–15 chest compressions are used when performing chest compressions simultaneously.
- The amount of gas delivered (tidal volume) should approximate 15 ml/kg at a maximum peak inspiratory pressure of 20–25 cmH₂O. This volume generally results in visible but minimal expansion of the chest or movement of the abdomen.
The lungs of cats and neonates, and alveoli in patients with restrictive types of lung disease (e.g. pneumonia, adult respiratory distress syndrome (ARDS), pulmonary fibrosis, diaphragmatic hernia) are easily overinflated. Lung overexpansion can lead to pulmonary barotrauma, inflammation, haemorrhage and pneumothorax. Smaller tidal volumes (6–10 ml/kg) at higher respiratory rates (15–20 breaths per minute) should be used in these patients.

Mouth-to-nose ventilation is performed if ET tubes and ventilatory assist devices are not available. It is accomplished by cupping both hands around the animal’s muzzle, placing the operator’s mouth against the thumbs (attempting to produce an airtight seal) and blowing air into the animal’s nose. Inflation of the stomach with air may occur, but can be avoided by pushing the larynx dorsally in order to occlude the oesophagus. This technique is inefficient, but easily performed and can be life-saving in puppies and kittens or in larger dogs and cats in acute respiratory distress situations after removal of foreign material from the upper airway.

Ventilatory support should not be stopped once signs of spontaneous ventilation begin. Intermittent positive pressure ventilation is usually necessary until the patient regains consciousness. Listening to the end of the ET tube, watching for normal ventilatory movement of the chest wall and continued monitoring of mucous membrane colour and capillary refill time will help to assess the adequacy of ventilation. Measurement of arterial pH and blood gases on a point-of-care analyser, non-invasive assessment of oxygen saturation using pulse oximetry ($S_pO_2 >90\%$) and end-tidal capnography are invaluable monitoring techniques if available.

Following extubation, nasal oxygen and oxygen cages help to maintain arterial oxygenation in conscious animals.

**Circulation**

**Closed-chest compression**

- Effective chest compression in small animals is accomplished by compressing the chest wall from side to side.
- Blood moves through the heart and vessels during chest compression, due to direct cardiac compression in cats and narrow-chested dogs, and/or to phasic increases in intrathoracic pressure, which collapses intrathoracic veins, in larger animals.
- Blood flow to the brain and heart is more effectively maintained by administering chest compressions simultaneously with ventilation at relatively high airway pressures.
1. Place the animal in lateral recumbency.
2. Use the heel of one hand to compress one side of the chest wall, while placing the palm of the other hand or a sand-filled pillow under the opposing chest wall for support. The thumb and forefinger can be used to accomplish the same manoeuvre in cats or in very small dogs.

3. Enough force must be generated to produce an obvious indentation of the chest wall of approximately 1–3 cm, depending on patient size.
4. A breath should be given once every fifth or sixth chest compression, or 2–3 times for every 10–15 chest compressions.
5. The ideal chest compression rate in dogs and cats is approximately **100 compressions per minute**, devoting equal time to compression and relaxation.

Intermittent slow abdominal compression (counterpressure) appears to be another effective means of improving blood flow during resuscitative efforts. Abdominal counterpressure is accomplished by having a second person use the palms of both hands to compress the abdominal cavity slowly at approximately 20-second intervals. Alternatively, the abdomen may be temporarily bound during CPCR with an elastic bandage or towel if extra help is not available.

6. Signs of restoration of effective peripheral blood flow include: improvement in mucous membrane colour; decrease in capillary refill time; reduction in pupil size; and restoration of a peripheral arterial pulse. The peripheral arterial pulse should be evaluated during a short pause in chest compression because pulse waves carried into the femoral veins during chest compression may be mistaken for an arterial pulse.

7. Chest compression is stopped when signs of a spontaneous return in cardiac output are noted.

8. Evaluate within 3–4 minutes. If signs of successful resuscitation are not evident, or if the patient continues to deteriorate, proceed to open-chest cardiac compression (see below).

**Open-chest cardiac compression**

- Direct cardiac compression always produces better blood flow than closed-chest compression and has been associated with improved survival and neurological outcome.
- However, it requires surgical intervention, predisposes to infection and has a prolonged recovery period.

*When to use:*
- If signs of successful resuscitation are not evident after 3–4 minutes of chest compression.
- If the patient continues to deteriorate (bradycardia or cardiac arrest).
- In patients with severe chest trauma, fractured ribs, pneumothorax, haemothorax, pericardial effusion, diaphragmatic hernia and other primary thoracic diseases (e.g. neoplasms, foreign bodies).

*Technique:*

1. To limit contamination, the hair should be clipped and the area cleaned quickly with chlorhexidine or povidone–iodine, followed by surgical spirit prior to making the approach.
2. Open the chest by making an incision from the top of the scapula to approximately 4 cm from the sternum on the left thoracic wall, at the cranial aspect of the 5th rib (fourth intercostal space), avoiding the intercostal vessels and nerves. Care must be taken not to damage the lung.

3. Once the chest is entered, spread apart the 4th and 5th ribs, and reflect the lungs dorsally and caudally.

4. Grasp the pericardium and open it near the apex of the heart. Reflect the pericardium dorsally to expose the ventricles.

5. Small patients: grasp the heart between the thumb and forefinger and initiate direct cardiac massage at approximately 100 compressions per minute. Larger patients: hold the heart between the palm and fingers and use a reverse milking motion to compress the heart.

Avoid excessive force during direct cardiac compression, as it can result in severe cardiac trauma, cardiac dysrhythmias and ventricular fibrillation.

6. Stop when signs of a spontaneous return in cardiac output are noted.

The colour, tone and rhythm of the heart can be evaluated once the chest is open. The presence of adequate ventricular filling can be assessed, and a decision made whether additional fluid resuscitation is required. In larger dogs, the aorta can be compressed dorsally against the spine with the thumb of the opposite hand, which promotes blood flow to the heart and brain.

**Fluid therapy**

- If cardiac arrest is thought to be secondary to hypovolaemia, aggressive fluid resuscitation may be warranted, especially once a spontaneous heart beat has occurred.
- However, in the euvoelaemic patient large amounts of intravenous fluids may be detrimental, especially if lung disease is also present. Excessive fluid therapy could lead to right-sided pressure loading and significant impairment of myocardial blood flow in animals with poor cardiac function. Therefore only small amounts of intravenous fluid (1–2 ml/kg/h), if any, should be administered until cardiac function is restored.

**Drugs**

- Once basic life support has been initiated, drugs may be used to promote restoration of spontaneous circulation or to address the underlying cause of the arrest.
Procedures in Small Animal Practice

- Drugs should always be administered by bolus injection and followed by 1–3 ml of crystalloid flush through a peripheral vein.
- Drug administration should occur simultaneously with chest compression, if possible, since the chest compressions are responsible for blood flow and the delivery of the drug to the heart and brain.
- Intravenous drug administration is preferred, although intracardiac and intratracheal (endobronchial) routes can be used when venous access is not readily available.
  - The intracardiac drug dose is generally one half the intravenous dose.
  - The endobronchial dose is generally 2–3 times the recommended intravenous dose.
- Using the intracardiac route, drugs are deliberately injected into the lumen of the left ventricle in order to hasten their delivery to the coronary arteries and ventricular myocardium.

Intracardiac injections are easily performed during open-chest resuscitation, but are not recommended prior to opening the chest, since inadvertent pneumothorax, haemopericardium or intramyocardial injections can occur, leading to cardiac dysrhythmias.

- Adrenaline, atropine and lidocaine can all be administered by the endobronchial route. The drugs are diluted in saline to administer 1 ml/5 kg, and followed by two or three manual lung inflations. It can be helpful to pass a sterile red rubber or male dog urinary catheter down the ET tube to facilitate instillation of the drug into the lower airways.
- Sodium bicarbonate should never be administered by the endobronchial route.

Monitoring
- Proper post-resuscitation monitoring and therapy are as critical as the resuscitation period itself if the patient is to survive.
- At particular risk is the CNS, where ischaemia/reperfusion can lead to neuronal damage, cerebral oedema and increased intracranial pressure.
- Careful monitoring of CNS signs (mental status), heart rate and rhythm, packed cell volume, total protein, arterial blood gas and acid–base parameters (pH, $pO_2$, $pCO_2$), electrolytes ($Na^+$, $K^+$, $Ca^{2+}$), and urine production (1–2 ml/kg/h) is essential.

Further information on CPR is available in the BSAVA Manual of Canine and Feline Emergency and Critical Care.
Cardiorespiratory examination

Indications/Use
- Successful management of the animal with cardiorespiratory disease depends on accurate anatomical localization of disease and efficient diagnostic planning.
- Determination of the history of the complaint, assessment of the pattern of breathing, and careful examination and auscultation will assist in determining the site responsible for generation of cardiorespiratory complaints.
- Electrocardiography and blood pressure measurement also form part of the assessment.

Equipment
- Stethoscope
- Stopwatch or timer

Patient preparation and positioning
- Assessment should be performed on the conscious animal.
- The patient should be standing if possible; this is particularly important for cardiac auscultation.
- Animals should be kept calm throughout the examination.

Observation

Sternal recumbency, standing with elbows abducted and/or hyperventilating with the neck extended may indicate adoption of a position for relief of dyspnoea.

Body condition and shape
- In general, animals with airway-oriented diseases (tracheal collapse, chronic bronchitis, feline bronchial disease) will be in excellent systemic health and have a stable (or often increased) body condition score.
- Animals with parenchymal, pleural or cardiac disease with failure are more likely to be systemically debilitated or cachexic.
- Animals with congenital heart disease may have stunted growth.
- Obesity may be associated with lower airway disease in small breed dogs.
- Animals with chronic hyperpnoea can show barrel-chested changes.

Gait
- Cats with myocardial disease are at risk of systemic thromboembolism and may present with limb paresis.
Breathing pattern

• A visual appraisal of the respiratory pattern is made before the patient is stressed by an examination:
  – Restrictive respiratory diseases, such as pleural effusion and parenchymal disorders (pneumonia or oedema), cause rapid and shallow respiratory motions
  – Obstructive disorders, such as bronchitis, result in slow, deep breathing
  – Increased expiratory effort, prolonged expiratory time and abdominal effort on expiration should be considered suggestive of chronic lower airway disease.

Physical examination

Mucous membrane colour

• Assess the colour of the mucous membranes:
  – Normal mucous membrane colour is pink to pale pink
  – Pallor can indicate anaemia, but can also be associated with severe peripheral vasoconstriction in low-output states or conditions of elevated sympathetic tone
  – Congested or flushed mucous membranes occur in venous congestion (right-sided congestive heart failure) and polycythaemia
  – Cyanosis indicates an increased concentration of desaturated haemoglobin; ranges from slightly ‘dusky’ in mild cases to nearly navy blue in patients with severe hypoxaemia. Cyanosis is most often seen with severe hypoxaemia resulting from respiratory disease (including pulmonary oedema or pleural effusion with congestive heart failure) but can also be seen with a right-to-left congenital shunt
  – Injected mucous membranes suggest toxaemia or septicaemia
  – Cherry red mucous membranes are seen in carbon monoxide toxicity.

Capillary refill time (CRT)

1. Apply pressure to the gum with a clean finger and then release.
2. This will cause blanching of the area.
3. The area should return to its normal colour within 1–2 seconds.

• An increased CRT (>2 seconds) may indicate systolic failure, shock, dehydration or hypovolaemia.
• Patients in pain, with septic shock or with fever may demonstrate a decreased CRT (<1 second).

Jugular veins

• Visually inspect both jugular veins for distension and pulsation.
• In long-haired animals, it is possible to get an impression of jugular venous distension without clipping the hair if the coat is wetted with spirit.
Distension
• In the absence of a cranial mediastinal mass or obstruction of the cranial vena cava, distension of the jugular veins above the level of the right atrium (RA) indicates elevation of right atrial pressures.
• For milder degrees of elevation of right-sided pressures, pressure on the cranial abdomen may increase venous return to the right heart sufficiently to cause temporary jugular distension (‘hepatojugular reflux’). The distension resolves when the abdominal pressure is released.

Pulsation
• Jugular pulsation may be prominent with tricuspid regurgitation or when atrial contraction occurs against a closed tricuspid valve.

Peripheral pulse
• Suitable sites for monitoring the pulse include:
  – The femoral artery on the medial aspect of the femur
  – The digital artery on the palmar aspect of the carpus
  – The coccygeal artery on the ventral aspect of the base of the tail
  – The dorsal pedal artery just distal to the tarsus, between the second and third metatarsals.

  1. Locate the artery with the fingertips.
  2. Assess the pulse character, rate and rhythm.
  3. Compare the pulse rate with the heart rate, preferably by simultaneous auscultation, to determine the presence of any pulse deficit.

  • Weak pulses may reflect poor stroke volume.
  • Pulses may be absent in patients with systemic thromboembolism, although blood flow may return within a few hours to a few days following acute embolization.
Abdominal palpation
1. Assess the size of the liver:
   • Liver enlargement will accompany right-sided heart failure in dogs
   • The liver may be more palpable than usual in some cats with severe hyperpnoea and air trapping.
2. Assess the size of the kidneys:
   • Small, scarred kidneys may be associated with systemic hypertension
   • This should prompt blood pressure measurement.
3. Assess for the presence of abdominal effusion:
   • Small abdominal effusions may be appreciated as ‘slipperiness’ of the small intestines
   • Larger ascitic effusions are unmistakable on percussion of a fluid thrill.

Cardiac auscultation
Heart rate
• Heart rate can be helpful in differentiating cardiac from respiratory disease in dogs where respiratory disease is associated with elevated vagal tone, leading to an exaggerated respiratory sinus arrhythmia.
• Dogs with chronic heart failure are more likely to have increased sympathetic tone and an increased heart rate, although the degree of tachycardia may be modest.
• Care should be taken not to place too much reliance on heart rate in brachycephalic dogs with suspected heart failure, as concurrent airway obstruction may cause sufficient elevation in vagal tone that sinus arrhythmia persists despite overt heart failure.
• Cats do not appear to develop a consistent tachycardia with heart failure, and may be more likely than dogs to develop sinus bradycardia with life-threatening congestive failure signs.

Heart rhythm
• Normal dogs will have:
  – A regular heart rhythm
  – OR a sinus arrhythmia, where heart rate speeds up with inspiration and then slows with expiration.
• Cats normally have a regular heart rhythm.
• Bradyarrhythmias can be recognized by a slow heart rate.
• Atrial fibrillation sounds characteristically ‘chaotic’ on auscultation, but can still be confused with frequent atrial or ventricular premature beats.
Intensity of heart sounds

- Two heart sounds (S1 and S2) are normally heard in dogs and cats:
  - S1 occurs after closure of the atrioventricular valves, and is heard loudest at the apex of the heart
  - S2 occurs after closure of the aortic and pulmonic valves, and is heard loudest at the base of the heart.

![Heart sounds](image)

- High cardiac output states, cardiac enlargement, thin body condition and increased sympathetic tone can increase the intensity of heart sounds.
- Pericardial effusion, severe myocardial failure, obesity, space-occupying lesions and pleural effusion may decrease intensity.
- Intensity may vary from beat to beat with atrial fibrillation and ventricular tachycardia.

Additional heart sounds

- Additional heart sounds (S3 and S4) are associated with diastolic ventricular filling. These sounds are called gallop sounds and should not be audible in the normal dog or cat.
- Delayed ventricular relaxation can also cause an audible gallop, and may account for the presence of a gallop in some geriatric cats without obvious structural heart disease.
- In all other groups of cats and dogs, a gallop sound can be considered to be a specific indication of myocardial disease or heart failure.

Murmurs and clicks

- Murmurs are produced when blood flow becomes turbulent.
  - They are more likely with increased velocity of blood flow (e.g. with valve stenosis/insufficiency or increased sympathetic tone).
  - Murmurs are characterized according to their timing, character and intensity.

  - ‘Flow’ or physiological murmurs tend to be fairly quiet (<grade 2 or 3) and may vary in intensity.
  - They may occur with anaemia or can be normal in young animals.
• Systolic clicks are transient heart sounds associated with degenerative mitral valve disease.

**Examination of the thoracic cavity**

**Thoracic palpation**
1. Palpate the thorax for swellings, pain, rib fractures, subcutaneous emphysema or oedema.
2. Assess the strength of the apex beat using the fingers of one hand.
3. Determine the point of maximal intensity of the heart beat, using a stethoscope or fingers of one hand, to indicate any displacement by intrathoracic masses or effusion, and to detect any thrill (vibration of turbulence in blood flow).
4. Using both hands, gently compress the thoracic cavity to assess for reduced compliance of the cranial thoracic cage, which could indicate a cranial mediastinal mass.

**Tracheal palpation**
• Palpate the full length of the cervical trachea, using one hand, and observe for signs of sensitivity (usually indicated by a cough reflex).
• Tracheal sensitivity is a non-specific sign of airway irritation and is usually present in airway or parenchymal disease.
• In dogs and cats with chronic bronchitis, airway collapse or pneumonia, a *harsh cough* is usually elicited with tracheal palpation.
• A *soft cough* is more typical in dogs with heart failure.

**Thoracic auscultation**
1. Using a stethoscope, auscultate all lung fields on both sides of the thorax.
2. Note the presence, type and location of any abnormal lung sounds:
   • *Normal* sounds are termed bronchial, vesicular or bronchovesicular
   • *Crackles and wheezes* are examples of abnormal noises produced in diseased lungs
   • Determining the specific phase of the respiratory cycle in which abnormal lung sounds occur is important for categorizing the sound and determining the most likely pathology
   • *Absence* of lung sounds is also an important finding:
     – It typically reflects disease of the pleural space
     – Consolidation of a lung segment can also lead to an absence of lung sounds
     – Loss of lung sounds *dorsally* generally indicates air accumulation
     – Loss of lung sounds *ventrally* indicates a pleural effusion or space-occupying mass.
Thoracic percussion
1. Place the fingers of one hand flat against the thoracic cage.
2. Curve the fingers of the other hand gently and hold them rigid.
3. Use the fingers of the free hand to strike the fingers on the chest wall, causing production of sounds.

4. Examine all lung fields to detect localized differences in sound transmission.
   - Percussion causes vibration of the chest and intrathoracic structures, and the pitch reflects the underlying air-to-tissue ratio within the thorax.
   - Over the heart, a dull percussive sound is heard because of the presence of soft tissue that dampens the transmission of sound.
   - When the chest cavity is filled with fluid, or the lung is consolidated by disease, dull sounds are noted in the affected areas; whilst over air-filled lung structures, more resonant sounds are heard.
   - In an animal with pneumothorax or air trapping, sounds have increased resonance.

More detail on these procedures and interpretation of the results can be found in the *BSAVA Manual of Canine and Feline Cardiorespiratory Medicine.*
Cast application

Indications/Use

- Additional external support to supplement internal fixation for:
  - Arthrodesis of carpus/tarsus
  - Fractures of the distal limb
- Primary coaptation of selected long bone fractures:
  - The fracture should be relatively stable, e.g. greenstick fractures, interdigitating transverse fractures, or fracture of one member of a pair of bones
  - The fracture should be distal to the elbow and stifle and in general be non-articular
  - Closed fracture reduction with at least 50% reduction in two radiographic planes should be achieved
  - Healing by secondary bone union (i.e. callus formation) should be possible within 4–6 weeks; this is more likely in younger animals

Contraindications

- Soft tissue swelling
- Athletic or working animals

Equipment

- 25 mm wide adhesive tape
- Tongue depressor
- Stockinette (not absolutely essential)
- Cast padding
- Conforming gauze bandage
- Resin-impregnated fibreglass cast materials
- Outer protective bandage material, e.g. self-adhesive non-adherent bandage or adhesive bandage (optional)

Patient preparation and positioning

- The animal should be sedated heavily or anaesthetized.
- The haircoat should be clipped if it is likely to interfere with cast application and the limb should be clean and dry.
- The animal should be placed in lateral recumbency with the affected limb uppermost and supported in a weight-bearing position by an assistant.

Technique

1. Place two strips of adhesive tape (stirrups) on the distal limb on either the dorsal and palmar/plantar surfaces or the medial and lateral surfaces. These stirrups should extend beyond the tip of the toes and should be stuck to each other or to a tongue depressor. The assistant can now maintain the limb elevated away from the body by holding the stirrups.
2. Roll the stockinette up the limb and apply tension to eliminate creases.
3. Beginning distally, apply cast padding in a spiral fashion up the limb, overlapping by 50% on each turn. Two layers are generally indicated. Take particular care to ensure even padding over pressure points. Excessive padding about pressure points should be avoided and consideration should be given to increasing the padding in adjacent depressed regions to create a bandage of even diameter.

4. Apply conforming gauze to compact the cast padding, again overlapping turns by 50%.

5. Follow the manufacturers’ recommendations regarding wetting and handling of the cast material.

6. Apply the cast material over the bandage, again with a 50% overlap on each turn. Two or three layers of cast material are generally needed. Leave a 1–2 cm margin of cast padding exposed proximal to the cast. Increase tension as the cast is applied proximal to the elbow or stifle, to give a snug fit about the muscle masses and to prevent loosening. Do not make indentations in the cast material with fingers.

7. Once the cast has hardened, an oscillating saw may be used to trim excess casting material proximally and distally to prevent rubbing and to permit weight bearing, respectively.

8. Roll the stockinette and padding over the proximal edge of the cast and secure them to the cast with adhesive tape.

9. Peel apart the stirrups, twist them through 180 degrees and stick them to the distal cast. The pads and nails of the axial digits should remain exposed.

10. Medication with non-steroidal anti-inflammatory drugs is useful to limit soft tissue swelling and to provide analgesia. The requirement for ongoing treatment should be reassessed after 3–5 days.

Additional/Alternative techniques

The cast may be cut along its lateral and medial aspects with an oscillating saw and then bandaged together with strong adhesive tape. This facilitates removal and replacement of the cast to check for problems, but will affect some of the material properties of the cast and is not recommended by some surgeons.

Cast maintenance

- Written instructions should always be given out at discharge and owners must understand their responsibility in cast maintenance.
- Casts should be checked every 4 hours for the first 24 hours and then weekly by a veterinary surgeon; rapidly growing dogs and other high-risk patients may require more frequent assessment.
- Animals with a cast should have restricted exercise levels.
- The cast must be kept clean and dry. A plastic bag may be placed over the foot while the dog is walking outside. The plastic bag should be removed when the dog is indoors.
- Points to monitor for are:
  - Swelling of the toes or proximal limb
  - Toe discoloration and coolness
Skin abrasion about the toes or proximal cast
- Cast loosening
- Angular deformity
- Cast damage or breakage
- Discharge or foul odour
- Chewing at the cast
- Deterioration in weight-bearing function
- Signs of general ill health (inappetence, dullness, etc.).

These signs should prompt cast removal, assessment and replacement only if appropriate.

**Potential complications**
- Venous stasis
- Limb oedema
- Moist dermatitis
- Skin maceration under a wet bandage
- Wound contamination
- Pressure necrosis
- Pressure sores
- Cast loosening
- Deterioration in fracture apposition
- Fracture non-union, malunion or delayed union
- Joint stiffness or laxity
- Complications may occur more frequently in growing animals, chondrodystrophic breeds and obese dogs

**Cast removal**
- The timing of cast removal should be aided by radiography. In most instances radiographic evidence of bridging callus formation across fracture sites or arthrodesis sites is desired prior to cast removal.
- Although plaster shears can be used to remove most casting materials, an oscillating circular saw is more suitable.
- Bilateral incisions are made in the cast, taking care not to damage underlying tissue. The two halves are then prised apart using cast spreaders if available, and the underlying bandage materials are removed.
- After cast removal it is important that a regimen of progressively increasing controlled exercise is enforced. The goal is stimulation of callus remodelling without jeopardizing fracture/arthrodesis repair.

**Catheterization** see
- Intravenous catheter placement
- Urethral catheterization

**Central line placement** see
- Intravenous catheter placement – (b) jugular vein
Cerebrospinal fluid sampling – (a) cerebellomedullary cistern

Indications
- Suspected meningitis
- Infectious CNS disease
- Pyrexia of unknown origin

- CSF is more commonly collected from the cerebellomedullary cistern as it is less difficult, usually results in a larger sample volume and is typically associated with less iatrogenic blood contamination than collection from the lumbar cistern.
- In cases of focal CNS disease, CSF samples are more likely to be abnormal or representative of the CNS when they are collected caudal to the lesion. Therefore, in animals with lesions involving the spinal cord or canal, lumbar cistern samples are more consistently abnormal than samples collected at the cerebellomedullary cistern. In this situation it is preferable to collect CSF from both sites.

Contraindications
- Suspected increased intracranial pressure (e.g. progressive obtundation; papilloedema; miosis with responsive pupillary light reflex; intermittent extensor rigidity with opisthotonos)
- Suspected active intracranial haemorrhage or haemorrhagic diathesis
- Atlantoaxial luxation or other causes of cervical vertebral instability
- Infection of the soft tissues overlying the puncture site
- Evidence of very large intracranial space-occupying masses
- Severe hydrocephalus or severe cerebral oedema on MRI

Equipment
- As required for Aseptic preparation – (b) non-surgical procedures
- Spinal needle: 19–22 G; 1.5 to 2.5 inch
- 2 ml syringe
- EDTA and sterile plain collection tubes
- Sedimentation chamber

Patient preparation and positioning
- General anaesthesia is essential.
- The animal is placed in lateral recumbency, with the dorsum near the table’s edge.
- Flex the head to 90 degrees and have an assistant hold it in place.
Aseptic preparation – (a) non-surgical procedures is performed on a wide area at the base of the skull, from the occipital crest and including the wings of atlas.

Technique
1. Palpate a triangle of landmarks formed by the occipital protuberance and the most prominent points of the lateral wings of the atlas.
2. The location for needle insertion is on the dorsal midline, halfway between the wings of atlas and the occipital protuberance.

Needle insertion site (X). A = atlas. B = occipital protuberance
Caution should be employed with Cavalier King Charles Spaniels. Numbers of this breed are affected with Chiari-like malformations, and sampling from the cerebellomedullary cistern may lead to needle penetration of the cerebellum and brainstem. It is therefore advisable to obtain a lumbar cistern sample unless there is MRI evidence to show that the cerebellum is not caudally displaced.

3. Insert the spinal needle, with the bevel facing caudally, parallel to the table surface and parallel to the nose.
4. Once the skin is penetrated, remove the stylet.
5. Advance the needle very slowly (1–2 mm at a time) whilst watching for CSF appearing in the hub. If the needle hits bone while being advanced, it may be redirected cranially or caudally, moving the needle off the bone until the subarachnoid space is penetrated.
6. When the subarachnoid space has been entered, CSF will appear in the needle hub. If blood is seen in the needle hub, entry into a local blood vessel is likely and the sample will be less useful for cytological evaluation. Remove the needle and make a fresh attempt with a new needle.
7. Collect CSF by allowing it to drip passively from the hub into collecting vessels. Suction with a syringe should not be applied, as this usually results in haemorrhagic contamination of the sample.

8. When a minimum of 0.5 ml CSF has been collected, withdraw the needle in a single motion.
Sample handling

- CSF is hypotonic compared to serum; the cells within CSF rapidly swell and burst due to osmotic lysis soon after collection. For best results analysis should ideally be performed within 30 minutes of collection.
  - Cells can be concentrated using a homemade sedimentation chamber. The flanged end of a 2 ml syringe barrel (the end where the plunger would be inserted) is clamped to a clean microscope slide using bulldog clips after smearing vaseline or another occlusive lubricant around the base to form an air-tight seal. 0.25–0.5 ml of CSF is put into the chamber, left for 30 minutes, and the supernatant then pipetted off. The slide with adherent cells is then air-dried.
  - Alternatively, cells can be preserved by the addition of 30–50% by volume of the animal's own serum (i.e. 0.3–0.5 ml serum to 1.0 ml CSF). If this is done, it is important also to submit a CSF sample in a plain tube without the addition of serum to enable total protein measurement.
- CSF in the EDTA tube is submitted for cytological examination, total protein and total nucleated cell count.
- CSF in the sterile plain tube can be submitted for serology and bacteriological culture if necessary.

Potential complications

- Loss of airway patency due to patient positioning and use of unguarded ET tube
- Cerebral and/or cerebellar herniation due to elevation in intracranial pressure
- CNS haemorrhage
- Seizures
- Brainstem trauma due to needle damage
- Death

Cerebrospinal fluid sampling – (b) lumbar cistern

Indications

- Lesions of the spinal cord or canal
- Suspected meningitis
- Infectious CNS disease
- Pyrexia of unknown origin

- In cases of focal CNS disease, CSF samples are more likely to be abnormal or representative of the CNS when they are collected caudal to the lesion. Therefore, in animals with lesions involving the spinal cord or canal, lumbar CSF samples are more consistently abnormal than samples collected at the cerebellomedullary cistern. In this situation it is preferable to collect CSF from both sites.
Contraindications
- Suspected increased intracranial pressure (e.g. progressive obtundation; papilloedema; miosis with responsive pupillary light reflex; intermittent extensor rigidity with opisthotonos)
- Suspected active intracranial haemorrhage or haemorrhagic diathesis
- Luxation or vertebral instability of the caudal lumbar vertebrae
- Infection of the soft tissues overlying the puncture site
- Evidence of very large intracranial space-occupying masses
- Severe hydrocephalus or severe cerebral oedema on MRI

Equipment
As for Cerebrospinal fluid sampling – (a) cerebellomedullary cistern

Patient preparation and positioning
- General anaesthesia is essential.
- The animal is placed in lateral recumbency, with the dorsum near the table’s edge.
- Flex the lumbar spine and have an assistant hold it in place.
- Locate the appropriate intervertebral space:
  - Dogs: L4–L5 or, preferably, L5–L6
  - Cats: L6–L7
  This is done by palpating the ilial crests: the vertebral spinous process found immediately cranial to the ilial crests is that of L6.
- Aseptic preparation – (a) non-surgical procedures is performed over on area at least 5 cm wide.

Technique
1. The needle is positioned on the midline, just cranial to the appropriate vertebral spinous process, at an angle of 45 degrees to the skin, with the bevel facing caudally.
2. Once the skin is penetrated, advance the needle very slowly (1–2 mm at a time). The stylet may be left within the spinal needle.

3. When correctly positioned, the needle typically passes through or alongside the cauda equina/caudal spinal cord, which often elicits a tail or leg twitch.

4. Remove the stylet.

5. When the subarachnoid space has been entered, CSF will appear in the needle hub. If blood emerges from the needle hub, entry into a local blood vessel is likely and the sample will be less useful for cytological evaluation. Remove the needle and make a fresh attempt with a new needle.

6. Collect CSF by allowing it to drip passively from the hub into collecting vessels. Suction with a syringe should not be applied, as this usually results in haemorrhagic contamination of the sample.

7. When a minimum of 0.5 ml CSF has been collected, withdraw the needle in a single motion.

Sample handling
As for Cerebrospinal fluid sampling – (a) cerebellomedullary cistern.

Potential complications
- Cerebral and/or cerebellar herniation due to intracranial pressure change
- CNS haemorrhage
- Seizures
- Spinal cord trauma due to needle puncture

Clotting tests see
- Buccal mucosal bleeding time
- Platelet count
- Whole blood clotting time

Coagulation tests see
- Buccal mucosal bleeding time
- Platelet count
- Whole blood clotting time

Colonoscopy see
- Endoscopy of the gastrointestinal tract – (b) lower
Cranial draw test

Indications/Use
• To diagnose partial or complete rupture of the cranial cruciate ligament (CCL)
• Note: this test does not identify isolated rupture of the caudomedial band of the CCL
• Often used in association with the Tibial compression test

Contraindications
Periarticular fibrosis and meniscal injury, with the caudal horn of the medial meniscus wedged between the femoral condyle and tibial plateau, may prevent cranial draw in a CCL-deficient stifle

Patient preparation and positioning
• Can be performed in the conscious animal. However, if the patient is tense (due to pain or temperament) or if the CCL is only partially torn, sedation or general anaesthesia may be required.
• A conscious patient may be restrained in a standing position on three legs, with the affected limb held off the ground.
• Sedated or anaesthetized patients may be positioned in lateral recumbency, with the affected limb uppermost.

Technique
1. Grasp the distal femur in one hand, placing the thumb over the lateral fabella and the index finger on the patella.
2. Use the other hand to grasp the proximal tibia, placing the thumb over the head of the fabella and the index finger on the tibial crest.
3. Apply a cranial force to the tibia while the stifl e joint is held in full extension, and then while the joint is held in 30–60 degrees of flexion.

Results

- Complete rupture of the CCL is associated with cranial displacement of the tibia relative to the femur, in both extension and flexion.
- Isolated rupture of the craniomedial band of the CCL is associated with cranial displacement of the tibia relative to the femur, in flexion only.
- A short cranial draw motion, with a sharp end point, may be detected in young animals and is normal.

More detail on this procedure and interpretation of the results can be found in the BSAVA Manual of Canine and Feline Musculoskeletal Disorders.

Cystocentesis

Indications

- Collection of urine without contamination from the urethra or genital tract, e.g. for bacteriological culture
- Decompression of a severely overdistended bladder pending urethral catheterization or, in exceptional circumstances, when urethral catheterization is not possible

Contraindications

- Severely diseased bladder (rupture possible)
**Equipment**
- As required for *Aseptic preparation – (a) non-surgical procedures*
- Hypodermic needles:
  - Dogs: 21–23 G, 1–2 inch
  - Cats: 23 G, 1–2 inch
- 5 or 10 ml syringe
- Sterile plain collection tube capable of holding at least 5 ml fluid
- Container with boric acid preservative

**Patient preparation and positioning**
- Cystocentesis can usually be performed with the patient under physical restraint or light sedation.
- In cats and small dogs, it is most readily performed with the animal in dorsal recumbency.
- In larger dogs, it may be performed with the animal standing or in lateral recumbency.
- *Aseptic preparation – (a) non-surgical procedures* is performed over the appropriate area.

**PRACTICAL TIP**
The bladder must contain a reasonable volume of urine, such that it can be safely identified and immobilized. If a small volume of urine is present or the animal is obese, ultrasonography may be used to help guide the needle into the bladder.

**Technique**
1. With one hand palpate and stabilize the bladder by pushing it in a caudal direction against the pelvic brim.
2. Attach the needle to a 5 or 10 ml syringe and insert through the abdominal wall, on the midline, just in front of the pelvic brim.
3. The ideal site of bladder penetration is a short distance cranial to the junction of the bladder with the urethra. Insert the needle in a caudal direction, at a 45-degree angle to the bladder wall if possible. Slight negative pressure should be applied to the syringe while inserting the needle.

*Dorsal recumbency*
*Alternatively*, in the standing dog, the sample can be obtained from the right side (to avoid penetrating the descending colon), while gently pushing the bladder from the left side towards the right side of the caudal abdomen.

4. Once the bladder lumen is penetrated, urine will be seen filling the syringe.
5. Once sufficient urine is collected to perform the required diagnostic tests (usually a minimum of 2 ml), remove the needle in one motion.

**Sample handling**
- Urine should be placed into a sterile plain tube for bacterial culture. Fresh urine should be cultured within 2 hours or may be refrigerated for up to 6 hours. Urine samples in boric acid preservative will keep for up to 72 hours.
- A separate sample in a plain tube can be used for *urinalysis*.

**Potential complications**
- Bladder rupture is rare but more likely if the bladder is diseased or the animal is not adequately restrained
- Uroperitoneum, following urine leaking from the needle site
Dexamethasone suppression test – (a) low dose

**Indications/use**
- Aid to the diagnosis of hyperadrenocorticism: reliably identifies the majority of adrenal-dependent cases and 90–95% of dogs with pituitary-dependent hyperadrenocorticism

**Equipment**
- As for Blood sampling – (b) venous
- Dexamethasone suitable for intravenous administration
- Intravenous catheter

**Patient preparation and positioning**
- This procedure should be carried out in the conscious animal, using manual restraint.
- Positioning and skin preparation are as for Blood sampling – (b) venous.
- The patient should be hospitalized for the duration of the test; avoid excessive exercise or stress.

**Technique**
1. Collect a blood sample (approximately 2 ml) from the jugular vein and place it into a heparin or plain collection tube to enable measurement of the basal cortisol concentration.
2. Inject 0.01 mg/kg of dexamethasone into the cephalic vein.

When dealing with a very small dose of dexamethasone, it is preferable to place an intravenous catheter to ensure that the entire dose is administered.

3. After 3 hours, collect a blood sample (approximately 2 ml) from the jugular vein and place into a heparin or plain tube. Label the tube clearly as the +3 hours sample.
4. After 8 hours, collect a further blood sample (approximately 2 ml) from the jugular vein and place into a heparin or plain tube. Label the tube clearly as the +8 hours sample.
5. Separate the serum or plasma prior to sending the samples to the laboratory.

For interpretation of results see the BSAVA Manual of Canine and Feline Endocrinology.
Dexamethasone suppression test – (b) high dose

Indications/Use
• Although no longer considered reliable, may be indicated in cases where the diagnosis of canine hyperadrenocorticism has been established by a screening test, but the differentiation of adrenal-dependent and pituitary-dependent hyperadrenocorticism has not been determined by ACTH measurement or diagnostic imaging
• Also used to aid diagnosis of feline hyperadrenocorticism, especially when combined with an ACTH response test

Equipment
As for Dexamethasone suppression test – (a) low dose

Patient preparation and positioning
As for Dexamethasone suppression test – (a) low dose.

Technique
1. Collect a blood sample (approximately 2 ml) from the jugular vein and place it into a heparin or plain tube to enable measurement of the basal cortisol concentration.
2. Inject 0.1 mg/kg of dexamethasone into the cephalic vein. When dealing with a very small dose of dexamethasone, it is preferable to place an intravenous catheter to ensure that the entire dose is administered.
3. After 3 hours, collect a blood sample (approximately 2 ml) from the jugular vein and place into a heparin or plain tube. Label the tube clearly as the +3 hours sample.
4. After 8 hours, collect a further blood sample (approximately 2 ml) from the jugular vein and place into a heparin or plain tube. Label the tube clearly as the +8 hours sample.
5. Separate the serum or plasma prior to sending the samples to the laboratory.

For interpretation of results see the BSAVA Manual of Canine and Feline Endocrinology.
Diagnostic peritoneal lavage

Indications/Use
- Where abdominocentesis has not yielded fluid, but suspicion for the presence of abdominal fluid or inflammation remains high
- It should only be performed after repeat abdominocentesis or ultrasound-guided aspiration

Contraindications
- Severe coagulopathy
- Marked distension of an abdominal viscus
- Marked organomegaly
- Suspected abdominal neoplasia

Equipment
- As required for Aseptic preparation – (a) non-surgical procedures
- A large bore 10–14 G over-the-needle catheter. The cannula can be fenestrated whilst on the metal stylet:
  - Use V-shaped incisions to create fenestrations in a spiral pattern around the catheter
  - Do not make fenestrations directly opposite each other (which would weaken the catheter)
  - No burrs must remain, as this could impair entry and exit of the catheter
  - Holes should not extend beyond 50% of the circumference of the catheter to avoid risk of breakage
- Local anaesthetic
- No. 11 or 15 scalpel
- 500 ml pre-warmed (to approximately 37°C) 0.9% sterile saline or lactated Ringer's solution (Hartmann's)
- Extension tubing
- 3-way tap
- 60 ml syringes
- 2 ml syringe
- EDTA and sterile plain collection tubes
- Microscope slides

Patient preparation and positioning
- In most cases, sedation is not needed and the patient is restrained manually in order to minimize movement and avoid accidental bowel puncture.
- The patient is restrained in lateral recumbency.
- Aseptic preparation – (a) non-surgical procedures is carried out on an area approximately 10 cm x 10 cm, centred on the umbilicus, and a fenestrated drape placed.

Technique
1. The site for diagnostic peritoneal lavage is a point approximately 1 cm below the midline and 1–2 cm caudal to the umbilicus. The skin and subcutaneous tissue may be infiltrated with local anaesthetic, if required.
2. Using a scalpel, make a small stab incision through the skin.
3. Introduce the fenestrated catheter and aim it caudally towards the pelvis. Once the abdominal wall is penetrated, remove the stylet, leaving the cannula in place.
4. Attach a 3-way tap and extension tubing.
5. Infuse warm saline into the abdomen via gravity flow or gentle injection. A total volume of 20 ml/kg bodyweight is infused.
6. Gently roll the animal from side to side to distribute the fluid, preferably with the catheter still in situ.
7. Remove the 3-way tap and allow the fluid to drain into the collection tubes.
8. If no fluid is obtained, attempt gentle aspiration with a 2 ml syringe. Note: Only a very small portion of the infused volume will be retrieved (usually only 1–2 ml); any remaining fluid will be absorbed across the peritoneal membrane.

Sample handling
- Place fluid in an EDTA tube for cytology. Note that a total nucleated cell count cannot be performed, as the dilution factor is unknown.
- Place fluid in a plain tube for total protein measurement and other biochemical-serological tests.
- A sample in a sterile plain tube can be submitted for bacteriological culture if necessary.
- Make several fresh air-dried smears (unstained).

Potential complications
- If blood is aspirated, stop. Place the blood in a glass tube and observe for clot formation. Blood from the abdominal cavity will not clot, whereas blood from a vessel or organ will clot. If bleeding persists, abdominal pressure should be applied via manual compression or a pressure bandage.
- If fluid is obtained that suggests that the gastrointestinal tract has been punctured, any hole should seal when the needle is removed. The patient should, however, be monitored for developing peritonitis.
- In some animals with large abdominal effusions, the centesis hole may continue to drain fluid. If this occurs, a pressure dressing should be applied for several hours.
- Extension of localized peritonitis.
- Dissemination of neoplastic cells.

Details of the cytological evaluation of peritoneal fluid samples are given in the BSAVA Manual of Canine and Feline Clinical Pathology.
Doppler blood pressure measurement  see
  • Blood pressure measurement – (b) indirect

DPL  see
  • Diagnostic peritoneal lavage

Duodenoscopy  see
  • Endoscopy of the gastrointestinal tract – (a) upper
Ehmer sling

Indications/Use
- To support the hip joint following closed reduction of hip joint luxation (to permit stabilization and healing of the periarticular tissues)
- The Ehmer sling holds the pelvic limb in flexion, while internally rotating and mildly abducting the hip joint

Contraindications
- Patient’s temperament will not tolerate prolonged immobilization of limb
- Patient’s conformation (e.g. chondrodystrophoid, well muscled) does not permit effective sling application
- Hip instability following closed reduction of hip luxation, such that the Ehmer sling would not hold the hip in the reduced position
- Concurrent hip fractures or pre-existing disease (e.g. hip dysplasia)
- Concurrent injuries such as fractures or open wounds

Equipment
- Padded bandage material or cast padding
- Conforming gauze bandage
- Adhesive outer protective bandage material (e.g. ‘Elastoplast’)

Patient preparation and positioning
- General anaesthesia is required for closed reduction of hip luxation and is preferred for Ehmer sling application.
- The patient should be positioned in lateral recumbency with the affected limb uppermost.
- At the beginning of the procedure the pelvic limb should be held in a gently flexed position.

Technique
1. Confirm successful hip joint reduction by radiography and check range of motion and stability of the hip joint. Confirm that conservative management of hip luxation is appropriate (see Hip luxation – closed reduction).
2. Apply padded bandage material around the metatarsal region to lightly pad this area.
3. Secure the padded bandage material around the metatarsal region with conforming gauze bandage. It is important to pass the conforming gauze bandage from lateral to medial around the dorsal aspect of the metatarsus and from medial to lateral around the plantar aspect of the metatarsus.

4. Completely flex the whole limb.

5. Pass the conforming gauze bandage from the lateral aspect of the metatarsus across the cranial aspect of the mid-tibia, to the medial aspects of the mid-tibia and mid-femur.

6. Pass the bandage over the cranial aspect of the mid-femur to the lateral aspect of the mid-femur.

7. Pass the bandage caudal to the stifle to the medial aspects of the distal tibia and proximal metatarsal region, before returning to the plantar aspect of the metatarsus.

8. The result should be a figure-of-8 bandage.

9. Apply a few more layers of conforming gauze bandage to secure the pelvic limb in the flexed position.

10. Apply an adhesive protective bandage material over the conforming bandage to overhang the edge of the conforming gauze to secure the bandage to the skin/fur.
Alternative techniques

• Ehmer slings are prone to slipping, especially over the cranial aspect of the stifle. These slings can be made using an elastic adhesive bandage alone, with some metatarsal padding. This decreases slipping but the sling is more difficult to remove, and skin irritation is more likely.
• The sling can also be secured around the abdomen by extending the bandage from the plantarolateral aspect of the metatarsus, lateral to the flexed pelvic limb and over the dorsal aspect of the body, before wrapping it around the abdomen.

Sling maintenance

• Recommendations for the duration of time for which Ehmer slings need to be maintained to prevent hip reluxation vary: 7 to 10 days is frequently quoted.
• Ehmer slings must be checked several times daily for complications: ideally every 4 hours.
• Exercise restriction must be enforced.

Potential complications

• If applied too tightly, potential complications include:
  – Paw swelling due to venous stasis
  – Irritation of the skin, especially over the medial thigh
  – Sloughing of the metatarsal pad
  – Pressure necrosis of the soft tissues
• If the sling becomes wet, moist dermatitis and soft tissue maceration may occur
• Ehmer sling loosening or slipping over the stifle are frequent problems
• Hip re-luxation can also occur due to inappropriate case selection or incorrect technique

Elbow luxation – closed reduction

Indications/Use

• Traumatic elbow luxation in the adult dog in the absence of fractures and congenital skeletal deformities
• Closed reduction should be attempted as soon as possible before development of an organized intra-articular haematoma

Contraindications

• Elbow luxation associated with fractures and/or avulsion of ligaments

Equipment

Radiography equipment
Patient preparation and positioning

- General anaesthesia is required.
- Confirm complete traumatic elbow luxation and rule out associated fractures by palpation and radiography.
- The patient is positioned in lateral recumbency with the affected limb uppermost.

Technique

1. Hold the elbow in complete flexion for a few seconds to fatigue the surrounding muscles.
2. Identify the radial head, semilunar notch of the ulna and humeral condyles by palpation.
3. Place the elbow in about 110 degrees of flexion. With the carpus flexed to 90 degrees, rotate the antebrachium inward (pronation) using the metacarpal region as a handle. With the thumb of the opposite hand, manipulate the anconeal process toward the lateral epicondyle of the humerus.
4. Apply medial pressure to the olecranon to force the anconeal process medial to the lateral epicondyle. Once this has been achieved, extend the elbow slightly to lock the anconeal process in this position.

5. Apply medial pressure to the radial head and increase the amount of inward antebrachial rotation. Gradually flex the elbow and adduct the antebrachium simultaneously to force the radial head medially, using the anconeal process as a fulcrum. Slight abduction of the elbow joint may help.

6. Successful reduction is usually accompanied by a dramatic return to a normal range of joint motion and restoration of normal anatomical relationships.
7. Confirm reduction by radiography.
8. Check the collateral ligaments for damage. Flex both the elbow and carpus to 90 degrees and check the maximum degree of lateral and medial rotation of the paw. When the collateral ligaments are intact, about 45 degrees of lateral rotation and about 90 degrees of medial rotation are possible. If the lateral or
medial collateral ligaments are damaged, the possible degree of lateral or medial rotation, respectively, doubles.

9. Support the elbow in extension. Prolonged immobilization can be associated with loss of range of motion but some degree of immobilization is required for healing of periarticular structures.
   - If the elbow is very stable after reduction, a Robert Jones soft padded bandage for 5–10 days, followed by 2 weeks of exercise restriction combined with passive flexion and extension of the elbow joint, have been recommended.
   - If the elbow is unstable after reduction but complete collateral ligament rupture is not suspected, a Robert Jones soft padded bandage or Spica splint for 2 weeks, followed by 3–4 weeks of exercise restriction combined with passive flexion and extension of the elbow joint, have been recommended.

Potential complications
- Re-luxation of the elbow joint
- Decreased range of motion
- Progressive osteoarthritis

More information on joint problems and their treatment can be found in the BSAVA Manual of Canine and Feline Musculoskeletal Disorders.

Electrocardiography

Indications/Use
- Evaluation of cardiac anatomical changes, arrhythmias and pericardial and pleural diseases
- Evaluation of cardiac therapy
- Investigation of exercise intolerance, dyspnoea, coughing, weakness, collapse
- Evaluation of trauma cases
- Suspected electrolyte abnormality, particularly hyperkalaemia
- Monitoring during anaesthesia or in critical care setting

Equipment
- Electrocardiograph
- Limb leads x4
- Crocodile clips with teeth filed down or adhesive ECG pads
- Adhesive tape
- ECG gel
- 70% surgical spirit
Patient preparation and positioning

- The animal should be calm and relaxed, before being placed on a surface that is electrically insulated, such as a rubber mat or thick blanket.
- The conventional position is right lateral recumbency, with the fore- and hindlimbs as perpendicular to the long axis of the body as is possible.
- In dyspnoeic or uncooperative patients, sternal recumbency, sitting or standing positions are acceptable. Complex amplitude and morphology are more variable in these non-standard positions, especially in dogs; however, improved patient compliance (in cats) may produce a better quality trace with fewer artefacts.
- Chemical restraint may cause changes in rhythm, but can be used as a last resort if the alternative is an uninterpretable trace.
- It is not necessary to remove hair before the clips are attached.
- Adhesive electrodes are applied directly to the pads without previous preparation.

Technique

ECG leads

- The electrodes may be colour-coded and marked as for human use. They are attached to the limbs as follows:
  - RED: marked RA; right forelimb
  - YELLOW: marked LA; left forelimb
  - GREEN: marked LL or F; left hindlimb
  - BLACK: marked N; right hindlimb.
- For routine short ECG, the electrodes are usually attached to the animal using crocodile clips. The clips are generally placed on the skin overlying bony protuberances, to minimize the effect of muscle interference. They are then sprayed with surgical spirit or covered in ECG gel to achieve good electrical conductivity.
- Limb electrodes are usually positioned fairly near to the body to reduce movement artefact, using the relatively hairless areas of skin just behind both elbows and in front of the left stifle. Hindlimb electrodes may also be attached to the skin overlying the gastrocnemius tendon.
- The neutral electrode may be placed anywhere but is usually placed in front of the right stifle.
- For long-term ECG monitoring, adhesive electrodes stuck on to the pads are best; adhesive tape ensures that they do not become dislodged.

ECG machine controls

- Sensitivity control: allows the operator to vary the number of centimetres on the paper that are equivalent to 1 mV. Most traces are recorded at 1 cm/mV, but if the complexes are so tall that they cannot be accommodated, decreasing the sensitivity will give a readable trace.
• Paper speed control: allows the operator to choose how quickly the trace is run, usually either 25 or 50 mm/s. Slow running may be useful to save paper in a long recording if looking for intermittent arrhythmias. The trace should be run at a fast paper speed for an animal with a fast heart rate or tachyarrhythmia.

• Filter: allows artefacts to be suppressed, evening out the trace. Its use often causes a marked decrease in the height of the QRS complexes, and this should be taken into account when the trace is interpreted.

• Lead selector: manual lead selection is preferred and the operator should select the six frontal plane leads (Lead I, II, III, aVR, aVL and aVF) in turn.

Recording
1. Once the patient is positioned and the leads attached, switch on the unit and check the sensitivity and paper speed.
2. Ensure that the sensitivity (vertical axis scale) is set to optimize complex size to a height that is clearly seen, without overlapping of leads, and fits the paper.
3. The filter should be turned off.
4. Record a strip of all six frontal plane leads at 25 or 50 mm/s, together with a prolonged Lead II ‘rhythm strip’ recorded at 25 mm/s.
5. Longer tracings should be recorded if arrhythmias are evident or intermittent.
6. Three to four minutes of trace is usually sufficiently sensitive for most intermittent arrhythmias.
Reference ranges

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<thead>
<tr>
<th>Parameter</th>
<th>Unit</th>
<th>Dogs</th>
<th>Cats</th>
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<tr>
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<td>120–240</td>
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<td>up to 220 for puppies</td>
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<tr>
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<td>Mean electrical axis (MEA)</td>
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<td>0 to +160</td>
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Further details of ECG interpretation are given in the BSAVA Manual of Canine and Feline Cardiorespiratory Medicine.

Endoscopy – respiratory tract see
- Bronchoscopy
- Endotracheal wash
- Rhinoscopy

Endoscopy of the gastrointestinal tract – (a) upper

Indications/Use
- Investigation of clinical signs of oesophageal, gastric and duodenal disease
- Investigation of radiographic abnormalities of the upper gastrointestinal tract
- Collecting samples from the upper gastrointestinal tract
Contraindications
• Suspicion of oesophageal perforation
• Inadequate investigation prior to endoscopy

Equipment
• Flexible endoscope: diameter 7–9 mm; insertion tube at least 1 m long; 4-way tip deflection; minimum 2.2 mm biopsy channel
• Endoscopic viewing equipment
• Facilities for suction and washing the lens
• Flexible endoscopic biopsy forceps
• Mouth gag
• Water-soluble lubricant (e.g. K-Y jelly)
• Container with 10% buffered formalin
• Hypodermic needle: 21 G
• Tissue cassette with foam insert (‘cell-safe’ frames) see Rhinoscopy

Patient preparation and positioning

Do not perform barium studies during the 24 hours prior to upper gastrointestinal endoscopy.

• Food must be withdrawn for at least 12 hours prior to the procedure.
• Water should be withheld for 1 hour prior to the procedure.
• If delayed gastric emptying is suspected, take a plain lateral radiograph prior to anaesthesia to ensure the stomach is empty.
• General anaesthesia is essential. The endotracheal (ET) tube should be tied to the mandible, not the maxilla. Avoid atropine if possible, as it affects both motility and secretions of the gastrointestinal tract.
• Place a mouth gag to avoid damage to the endoscope.
• The patient should be positioned in left lateral recumbency, with the head and neck extended.

Technique
1. Lubricate the tip of the endoscope with K-Y jelly, avoiding the lens.
2. Pass the endoscope along the midline of the hard palate into the pharynx, reaching the upper oesophageal sphincter.
3. Apply gentle pressure to pass the sphincter.
4. Advance the endoscope in short segments down the oesophagus, whilst trying to keep the entire mucosal circumference in view and whilst insufflating with air.
5. Pass the endoscope until the gastro-oesophageal junction is visualized, usually as a star- or slit-like opening.
6. Align the tip of the endoscope with the lower oesophageal sphincter and gently advance, overcoming slight resistance.
7. If the lower oesophageal sphincter is closed, slightly angle the tip (30 degrees), with continued insufflation, to pass into the stomach.
8. Once inside the **stomach**, inflate with moderate amounts of air to allow orientation. The initial view on entering the stomach is of the junction of the fundus and the body on the greater curvature.

9. If duodenoscopy is to be performed, the stomach should be examined *after* examination of the **duodenum**. Delay in intubating the pylorus can make pyloric intubation more difficult.

10. Follow the rugal folds toward the antrum. Once in the antrum there are few rugal folds, the mucosa is paler in colour and the lumen tapers toward the pylorus. A ring of contraction passing toward the pylorus is also often present.

In larger dogs, as the endoscope approaches the antrum, there is a tendency either for the tip to get stuck in the greater curvature or, as the endoscope is advanced, the tip may appear to move backwards, i.e. away from the pylorus. This so-called paradoxical movement happens particularly when the stomach is over-inflated. It can be prevented by deflating the stomach as much as possible, or by turning the tip slightly into the greater curvature so that it advances into the antrum, or by rotating the endoscope slightly as it is advanced. Finally, if all else fails, external compression on the right body wall flattens the flexure and may assist entry into the antrum.

11. Position the tip of the endoscope against the pylorus. Maintain this pressure until the next antral contraction and the endoscope may be 'accepted' into the pylorus.

**Manoeuvres to aid pyloric intubation:**
- Deflate the stomach as much as possible
- Rotate the endoscope around its long axis
- Position the patient on its back or in right lateral recumbency. This is best done with the endoscope already positioned in the antrum, as orientation can be more difficult once the animal is repositioned
- With care, pass biopsy forceps ‘blindly’ through the pylorus and then pass the endoscope along this temporary ‘guide wire’.

12. ‘Red out’ occurs as the endoscope passes into the pyloric canal. The colour will change from ‘red out’ to ‘yellow out’, indicating the presence of bile in the duodenum.

13. As the endoscope is passed into the duodenum, angle the tip downwards and to the right.

14. Once in the duodenum, stop advancing the endoscope and inflate with air.

15. Once the duodenum has been examined, return the endoscope to the stomach.

16. Inflate the stomach to allow a complete examination.
17. Retroflex the endoscope (the so-called ‘J manoeuvre’) to allow visualization of the cardia, which lies in the ‘blind spot’ on entry to the stomach.

18. Deflate the stomach at the end of the procedure and withdraw the endoscope slowly, whilst examining the oesophagus.

Biopsy and sample handling

- Position the endoscope in the region to be sampled.
- Pass the biopsy forceps down the biopsy channel of the endoscope, with the cup firmly closed.
- Pass the forceps out of the end of the endoscope, open the cup, position on to the mucosa, and close the cup.
- Withdraw the biopsy forceps through the biopsy channel of the endoscope with the cup firmly closed.

**PRACTICAL TIPS**

- Position the biopsy forceps perpendicular to the mucosa whenever possible.
- Avoid over-inflation, as this stretches the mucosa and smaller samples will be obtained.

• Samples can be collected from:
  – The gastric fundus (take at least 2)
  – The body of the stomach (take at least 4)
  – The antral canal (take at least 2)
  – The periphery of any ulcer
  – Different areas of the duodenum (take 8–10).

If pyloric intubation is impossible, the duodenum can still be sampled, *with care*, by passing the forceps ‘blindly’ through the pylorus. However, it is *unsafe* to take repeated samples from the same site without being able to view it.

• Samples should always be taken, even if no lesions are apparent.
  • To remove the samples from the biopsy forceps, immerse in 10% buffered formalin. Then rinse the forceps in saline before reinserting them into the endoscope.
  • *Alternatively*, carefully remove samples from the biopsy forceps with a needle and place directly into formalin or lay on the foam insert of a tissue cassette.
Potential complications

- Gastrointestinal perforation is rare, but can result from forceful insertion of the endoscope, especially when attempting duodenal intubation. Excessive insufflation can also rupture ulcerated areas
- Significant haemorrhage is rare
- To avoid gastric dilatation, air should be removed from the stomach after gastroscopy
- Overdistension of the stomach causes compression of the caudal vena cava and a drop in venous return. Compression of the diaphragm may also result in decreased tidal volume
- Acute bradycardia most commonly occurs when the duodenum is entered, especially in toy breeds or in patients with severe gastrointestinal disease

Further information on endoscopy of the gastrointestinal tract can be found in the BSAVA Manual of Canine and Feline Endoscopy and Endosurgery.

Endoscopy of the gastrointestinal tract – (b) lower

Indications/Use

- Investigation of clinical signs of lower gastrointestinal tract disease
- Investigation of radiographic abnormalities of the lower gastrointestinal tract
- Collecting samples from the lower gastrointestinal tract

Contraindications

- Inadequate investigation prior to endoscopy

Equipment

- As for Endoscopy – (b) upper gastrointestinal tract (mouth gag not required)
- Alternatively, a rigid endoscope can be used, as this allows excellent visualization of the mucosa of the distal colon and rectum
- Laxative
- Equipment for giving an enema, e.g. Higginson’s pump

Patient preparation and positioning

- Food must be withdrawn for at least 24 hours prior to the procedure.
- Ideally, a laxative (e.g. Klean Prep <20 ml/kg orally or Dulcolax 5–20 mg/dog or 2–5 mg/cat orally) should be administered 24 hours prior to the procedure.
• Two warm water enemas should be given on the morning of the procedure, with the last one approximately 2 hours before the procedure. A maximum volume of 15 ml/kg should be administered, using a Higginson’s pump or equivalent. **A rectal examination must be performed first, to make sure it is safe to insert the enema tubing.**
• Water should be withheld for 1 hour prior to the procedure.
• If inadequate preparation is suspected, take a plain lateral radiograph prior to anaesthesia.
• **General anaesthesia is essential.** Atropine should be avoided if possible, as it affects both motility and secretions of the gastrointestinal tract.
• The patient should be positioned in left lateral recumbency.

**Technique**
1. Lightly lubricate the distal 20 cm of the endoscope, taking care to avoid the lens.
2. Insert the endoscope gently through the anus and into the rectum.
3. Once in the rectum, inflate with air to enable the mucosa to be visualized.

**PRACTICAL TIP**
Pinching the anus will prevent air escaping.

4. Pass the endoscope along the descending colon, while insufflating with air.
5. At the cranial end of the descending colon, the junction between the descending and transverse colons (splenic flexure) will be encountered as an obvious ‘bend’. The tip of the endoscope should be moved in the direction of the bend and advanced slowly. Some resistance may be felt, but excessive pressure should not be needed, and could result in rupture of the colon. It is not uncommon to induce ‘red-out’ whilst doing this.
6. Once in the transverse colon, an image of the mucosa should be re-established.

7. The next ‘bend’ marks the junction of the transverse and ascending colon; manoeuvre the endoscope in the direction of the bend and advance slowly, as before. Gently moving the endoscope backwards and forwards, while inflating with air, can aid its passage.

8. The ascending colon is short and ends at the ileocolic junction. This is recognized by the adjacent blind-ending sac, the caecum.

9. Carefully examine the caecum.

10. Withdraw the endoscope slowly and take biopsy samples. It is often easier to examine and sample the colon while the endoscope is being withdrawn.

**Biopsy and sample handling**

- Position the endoscope in the region to be sampled.
- Pass the biopsy forceps down the biopsy channel of the endoscope, with the cup firmly closed.
- Pass the forceps out of the end of the endoscope, open the cup, position on to the mucosa, and close the cup.
- Withdraw the biopsy forceps through the biopsy channel of the endoscope with the cup firmly closed.

**PRACTICAL TIPS**

- Position the biopsy forceps perpendicular to the mucosa whenever possible.
- Avoid over-inflation, as this stretches the mucosa and smaller samples will be obtained.
- Advance the forceps until the mucosa ‘tents’ and then close them.
• Samples can be collected from:
  – Ascending and transverse colons (take 2 or 3 from each)
  – Descending colon (take 4 or 5).
• **Samples should always be taken, even if no lesions are apparent.**
• To remove the samples from the biopsy forceps, immerse in 10% buffered formalin. Then rinse the forceps in saline before reinserting them into the endoscope.
• **Alternatively,** carefully remove samples from the biopsy forceps with a needle and place directly into formalin or lay on the foam insert of a tissue cassette.

**Potential complications**
• Haemorrhage (rare)
• Perforation (rare)

Further information on endoscopy of the gastrointestinal tract can be found in the *BSAVA Manual of Canine and Feline Endoscopy and Endosurgery.*

**Endotracheal wash**

**Indications/Use**
• To obtain a sample from the airways for cytology and bacteriology
• Generally yields samples representative of the trachea and primary or (at best) secondary bronchi, although some material from the lower bronchioles and alveoli may be collected

• The upper airway of medium-sized and large dogs may also be sampled by **transtracheal wash.**
• The lower airways of dogs and cats may be sampled using **bronchoalveolar lavage.**

**Contraindications**
• Compromised respiratory function

**Equipment**
• Sterile endotracheal (ET) tube
• Male dog urinary catheter (4–6 Fr). The tip of the catheter can be cut off to remove the side holes, but care must be taken to ensure that the tip is not sharp
• Topical local anaesthetic spray
• Warmed 0.9% sterile saline
• 10 ml and 20 ml syringes
• 3-way tap
• Sterile plain and EDTA collection tubes
• Microscope slides

Patient preparation and positioning
• General anaesthesia is essential.
• The patient is placed in either lateral or sternal recumbency.

Technique
1. The urinary catheter will be passed into the trachea through the ET tube. Mark the packaging of the urinary catheter to identify the depth of insertion into the ET tube, which will allow the catheter tip to extend 2–3 inches beyond the distal end of the ET tube.
2. If intubating a cat, apply topical local anaesthetic spray to the larynx.
3. Insert a sterile ET tube into the trachea, avoiding oral contamination.

Take care to minimize contact between the tip of the ET tube and the oropharynx during intubation, to avoid oropharyngeal contamination.

4. Pass the sterile catheter down the ET tube to the pre-marked length. To avoid contamination, the catheter should be inserted by feeding it through the sterile packaging.
5. Attach a 3-way tap to the catheter.
6. Inject warmed sterile saline (0.5 ml/kg) into the catheter via the 3-way tap.
7. Immediately aspirate back the saline.
8. Repeat the injection of saline and the aspiration two or three times as required. Coupage and turning the patient may improve cell yield.

Sample handling
• Submit a portion of the sample in a sterile plain tube for culture.
• Place an aliquot in an EDTA tube for cytology.
• Fresh air-dried smears of any flocculent/mucoid material can also be made and submitted to the laboratory for cytology.
Potential complications

- Larynx or airway spasm
- Catheter breakage and aspiration of the catheter into the airway
- Worsening of respiratory status due to stress of the procedure

Details on cytology of upper respiratory tract samples can be found in the *BSAVA Manual of Canine and Feline Clinical Pathology*.

Epilepsy  *see*

- Seizures – emergency protocol
Fine needle aspiration

Indications/Use
- To obtain a sample for cytology from soft tissue masses or abdominal viscera

Contraindications
- Coagulopathy

Equipment
- As required for Aseptic preparation – (a) non-surgical procedures
- Hypodermic needles: 21–23 G; ¾ to 3 inch (depending on depth of tissue to be sampled)
- 5 ml syringe
- Microscope slides
- Ultrasound equipment (where required)

Patient preparation and positioning
- Fine needle aspiration of superficial masses can usually be performed with the patient under physical restraint or light sedation.
- Fine needle aspiration of abdominal viscera or masses may require heavy sedation or general anaesthesia.
- The animal is positioned with the area to be sampled uppermost.
- Aseptic preparation – (a) non-surgical procedures is performed on the skin over the site to be aspirated, or the skin overlying the needle insertion site (for abdominal viscera/mass aspiration).

Technique

1. For superficial masses, immobilize the mass with one hand and insert the needle into the lesion using your other hand. For abdominal viscera/masses, immobilization is usually not possible.

2. Either
   Move the needle to and fro within the lesion several times and then remove it.
   Or
   For firm masses that are less likely to exfoliate, suction can be applied either continuously or intermittently, although this method can cause more damage to fragile cells and increase blood contamination.
To perform a continuous suction technique:
(i) Withdraw the plunger to one-half to three-quarters of the volume of the syringe (to apply 2–3 ml suction)
(ii) Maintain suction and move the needle to and fro, redirecting the needle several times within the lesion
(iii) Release the suction and remove the needle from the mass
(iv) Disconnect the needle from the syringe.

3. Attach an air-filled syringe to the needle.
4. Expel the contents of the needle containing the sample on to one or more clean microscope slides and prepare smears using one of the techniques described below.

**Smear preparation**

**Squash preparation**
1. Expel the aspirate on to the centre of a microscope slide.

![Image of microscope slide](image1)

2. Place a second spreader slide horizontally and at right angles to spread the sample, taking care not to exert too much downward pressure to avoid rupturing the cells.

![Image of spreader slide](image2)

3. Draw the spreader slide quickly and smoothly across the bottom slide. **Note that it is the smear produced on the underside of the spreader slide that is examined with the microscope.**
Blood film technique
This technique is useful for fluid aspirates (but not those with low cellularity). A ‘spreader’ slide with one corner missing is used to prepare the smear in order to avoid spreading the cells over the edges of the slide.

1. Expel the aspirate near to one end of a microscope slide in the centre line.
2. Hold the ‘spreader’ between the thumb and middle finger, placing the index finger on top of the ‘spreader’.
3. Place the ‘spreader’ in front of the aspirate, at an angle of about 30 degrees, and draw it backwards until it comes into contact with the sample, allowing it to spread out rapidly along the edge of the ‘spreader’.
4. Advance the ‘spreader’ forwards smoothly and quickly.
5. After the ‘spreader’ has been advanced two-thirds of the way along the bottom, lift it abruptly upwards in order to concentrate cells at the end of the smear.

Potential complications
- Minor haemorrhage. Any haemorrhage should be controlled by firm pressure over the site for several minutes. Continued haemorrhage from an internal organ is an indication for exploratory surgery
- Tissue damage

Cytology of FNA specimens is discussed in the BSAVA Manual of Canine and Feline Clinical Pathology.

Fluorescein test

Indications/Use
- Diagnosis of corneal ulcers, where epithelium is absent and the stroma is exposed
Contraindications
• Fluorescein will *not* stain healed ulcers that have re-epithelialized or the Descemet’s membrane

Equipment
• Fluorescein test strips or solution: *only disposable* sources, such as impregnated strips or single dose vials, should be used, as multi-dose bottles are readily colonized by *Pseudomonas aeruginosa*
• 0.9% sterile saline
• 2 ml syringe
• Cotton wool
• A blue light source, such as cobalt blue

Patient preparation and positioning
• This test is performed in the conscious animal without sedation.
• The animal should be restrained in a standing or sitting position.

Technique
1. Using a syringe, wet the test strip with sterile saline (this may not be necessary in patients with ample tear production).
2. Touch the test strip on to the dorsal or ventral bulbar conjunctiva. *Alternatively*, apply one drop of fluorescein solution to the eye.
3. Irrigate the eye with further saline to flush excess fluorescein from the ocular surface.
4. Examine the eye using a blue light source. Positive staining indicates a defect in the epithelium and, hence, an ulcer.

Further information on this technique and its interpretation is given in the *BSAVA Manual of Small Animal Ophthalmology*.

**Fits** see
• Seizures – emergency protocols

**FNA** see
• Fine needle aspiration
Gastric decompression – (a) orogastric intubation

Indications/Use
- Stabilization of dogs with gastric dilatation and volvulus (GDV) prior to surgery
- Orogastric intubation is superior to percutaneous needle decompression (see below)

Fluid therapy for the treatment of hypovolaemic shock must be initiated prior to gastric decompression.

Equipment
- Wide-bore stomach tube
- 7.5 cm wide roll of adhesive bandage with a hollow plastic core
- Funnel
- Bucket
- Warmed normal (0.9%) saline, lactated Ringer’s (Hartmann’s) solution or tap water

Patient preparation and positioning
- Sedation should be avoided, so as not to worsen hypotensive changes.
- A right lateral abdominal radiograph should be obtained to confirm the diagnosis of GDV if there is any doubt.
- The patient should be on a table or trolley to allow gravity to aid in gastric emptying.
- A ‘sitting’ position or sternal recumbency is often best, but gastric decompression may be achieved in right lateral recumbency.

Technique
1. Mark the distance from the dog’s nose to its 11th rib on the stomach tube. The tube should not be passed beyond this length, to minimize the risk of rupture of a potentially compromised gastric wall.
2. Insert the bandage roll longitudinally into the dog’s mouth. The mouth is held closed around the bandage roll by an assistant or by applying tape around the dog’s muzzle.
3. Insert the stomach tube through the core of the bandage roll and gently down into the dog’s stomach. Rotating the stomach tube gently about its long axis may aid passage through the gastro-oesophageal junction.
4. Gaseous decompression is achieved spontaneously.
Further emptying of the stomach can be achieved by lavage with copious volumes of warm saline, lactated Ringer’s or tap water.

a. Pour the solution from a height into the stomach, using a funnel attached to the end of the stomach tube.
b. Allow stomach contents to flow out of the stomach by lowering the tube below the level of the dog.

Potential complications
- Trauma to the gastro-oesophageal junction
- Passage of the stomach tube through a compromised gastric wall

Gastric decompression – (b) percutaneous needle

Indications/Use
- May facilitate orogastric intubation
- Should be performed when orogastric intubation (see above) is not possible

Equipment
- As for Aseptic preparation – (a) non-surgical procedures
- 16 or 18 G over-the-needle intravenous catheter

Patient preparation and positioning
- Sedation should be avoided, so as not to worsen hypotensive changes.
- A right lateral abdominal radiograph should be obtained to confirm the diagnosis of GDV if there is any doubt.
- The patient is placed in left lateral or sternal recumbency.
- Aseptic preparation – (a) non-surgical procedures of an area of skin over the most distended part of the right abdominal wall.

Technique
1. Percuss the right abdominal wall and define the site of greatest tympany, where the distended stomach is likely to be directly abutting the abdominal wall.
2. Insert the catheter percutaneously directly into the stomach.
3. Remove the stylet of the catheter to allow air to escape freely from the stomach.

Potential complications
- Entry of the catheter into another abdominal organ

Gastroscopy see
- Endoscopy of the gastrointestinal tract – (a) upper
Gastrostomy tube placement – (a) endoscopic

Indications/Use
Nutritional support:
- Long-term (months to years) in chronically ill animals
- Where naso-oesophageal or oesophagostomy tube feeding is not possible (e.g. vomiting, severe oesophageal disease)

Contraindications
- Gastric outflow obstruction
- Chronic vomiting
- Comatose, recumbent or dysphoric animals that are at risk of aspiration
- Very temporary anorexia

Equipment
- Mushroom-tipped catheter:
  - Cats: 18–20 Fr
  - Dogs: 18–24 Fr
- OR a commercial PEG (percutaneous endoscopic gastrostomy) tube kit for endoscopic placement
- Endoscope: 7–10 mm diameter; 1 m length
- Endoscopic grasping or biopsy forceps
- Hypodermic needle 12–14 G or over-the-needle catheter
- A piece of strong suture material, fishing line or wire long enough to extend from the mouth to the flank, with approximately another 30 cm to spare
- Disposable pipette tip or catheter tip of a 60 ml plastic syringe
- As required for Aseptic preparation – (a) non-surgical procedures
- No. 15 or 20 scalpel
- Non-absorbable suture material and needle
- Sterile dressing to cover the tube site
- Tube gauze or loose body wrap

Patient preparation and positioning
- General anaesthesia is required.
- The patient is positioned in right lateral recumbency.
- Perform aseptic preparation – (a) non-surgical procedures on an area on the left flank (approximately 15 cm x 15 cm in a medium-sized dog) extending caudally from the costal arch and dorsoventrally from the transverse processes of the lumbar vertebrae to the level of the ventral end of the 13th rib.

Endoscopic placement technique
1. Insert the endoscope into the stomach and insufflate the stomach as much as possible.
2. An assistant applies pressure with a finger into the dilated stomach just behind the last rib. This allows identification of the site into which the tube will be inserted. The endoscopist visualizes the indented mucosa made by the assistant's finger. Additionally, the light of the endoscope may be seen shining through the body wall and used to locate the site of catheter insertion.

3. A small skin incision is made over the optimal site for tube insertion. Ideally, the insertion point should be at the junction of the body and the antrum; if it is too near the pylorus the mushroom tip may cause an obstruction.

4. Withdraw the endoscope back into the cardia for safety.

5. Insert a needle or over-the-needle catheter percutaneously into the inflated stomach via the skin incision.

6. Pre-place open endoscopic forceps over the needle or catheter tip as soon as it enters the stomach.

7. Thread a line (suture material, fishing line or wire) through the needle or catheter into the stomach; then withdraw the needle or catheter.

8. Use endoscopic forceps to grasp the line and retract the endoscope and forceps slowly through the mouth, bringing the line with them.

9. If using a mushroom-tipped catheter:
   i. Cut a disposable pipette tip or catheter tip from a 60 ml syringe and thread it on to the line, with the conical point towards the mouth
   ii. Attach the line securely to the non-mushroom end of the gastrostomy tube
   iii. Transfix the line to the tube, using a needle inserted temporarily, before tying the knot
   iv. Tuck the end of the tube within the pipette tip.

If a commercial kit is being used, follow the manufacturer's instructions.
10. Pull the line entering the flank, to draw the feeding tube into the stomach and up to the body wall.

11. Apply strong traction to force the feeding tube through the body wall. It is usually necessary to make a small skin incision to ease its passage.

12. Once the tube has exited the body wall, pull it so that the mushroom tip lies snugly against the stomach wall. This should be checked endoscopically.

13. Secure the tube to the outside of the body with a Chinese finger-trap suture. Commercial kits often have devices to secure the tube in place.

14. Note the position of the tube against centimetre markers on the tube in case of future migration.

15. Cut off the end of the tube (eliminating the pipette tip or, in the case of a commercial kit, the swaged-on wire loop) and fit a syringe adapter and port so that the tube can be capped.

16. Apply a sterile dressing to the skin stoma site.

17. Place tube gauze or a loose body wrap to protect the remaining tube and prevent it from being removed.

**Tube care and maintenance**

- Inspect and clean the stoma site daily, applying antibiotic cream if necessary.
- The first gastrostomy tube can be left in place for at least 6 months.

**Feeding technique**

- Do not use the tube for the first 24 hours, to allow a primary seal to form between the stomach and body wall.
- Instil sterile saline initially, in case the tube has migrated. If there is any doubt as to its position, instil sterile iodinated contrast medium and take radiographs of the stomach.
- Before feeding each time, flush the tube with small amounts (5–10 ml) of lukewarm tap water.
Aspirate the contents of the stomach with an empty syringe prior to feeding. If gastric emptying is delayed and there is more than half the previous meal in the stomach, skip the feed and consider motility modifiers such as metoclopramide.

Warm food to body temperature and inject into the stomach over several minutes.

Liquid feeding is introduced, gradually increasing from one-third to two-thirds to the full daily calorific requirement (see **Resting energy requirement**) over 3 days.

Divide the daily requirement into multiple (4–6) feeds per 24 hours.

Flush the feeding tube after feeding, with 5–10 ml of lukewarm tap water.

The gastrostomy tube should remain in place for at least 7–10 days before removal, to allow permanent adhesions to form between the stomach and the body wall, and to prevent leakage of gastric contents into the peritoneal cavity.

If a gastrostomy tube is accidentally removed before intended, it can be replaced through the same stoma site if the procedure is performed rapidly (e.g. within 24 hours of the tube removal).

**Tube removal**

- In dogs >20 kg, cut the tube at the skin surface and allow the remaining internal portion to pass naturally through the gastrointestinal tract.
- In cats and small dogs (<20 kg), grasp the internal portion of the tube with endoscopic forceps. Cut the tube at the skin surface and pull the internal portion out through the mouth.

**Potential complications**

Major complications of feeding tube placement in dogs and cats are uncommon and can usually be avoided with proper technique and careful client counselling. The most common complications are:

- Tube blockage: flush after feeds with clean water. Carbonated drinks can be used to clear blocked tubes

- If the tube is not capped, leakage of gastric acid can cause acid burns on the skin

- Infection at the tube site

- Dislodgement of the tube by the animal

- Vomiting following feeding

Other complications include:

- Leakage around the tube site

- Diarrhoea due to overfeeding the small intestine

- Delayed gastric emptying due to effects of the tube on gastric motility

- Splenic laceration

- Necrosis of the gastric wall if the tube is too tight
Further details on placement and use of gastrostomy tubes can be found in the BSAVA Manual of Canine and Feline Gastroenterology, BSAVA Manual of Canine and Feline Endoscopy and Endosurgery and the BSAVA Manual of Canine and Feline Rehabilitation, Supportive and Palliative Care.

Gastrostomy tube placement – (b) surgical

Indications/Use
• Where a gastrostomy tube is required in a patient undergoing another surgical procedure

Equipment
• Mushroom-tipped catheter
  – Cats: 18–20 Fr
  – Dogs: 20–24 Fr
• Gastrostomy tube connector and bung
• As required for Aseptic preparation – (b) surgical procedures
• Suture materials
• Soft tissue surgical instrument set

Patient positioning and preparation
• General anaesthesia is required.
• The animal should be placed in right lateral recumbency or in dorsal recumbency.
• Aseptic preparation – (b) surgical procedures is required.

Technique
1. The stomach is approached via a left paracostal coeliotomy or a ventral midline coeliotomy.
2. Make a full thickness stab incision in the left abdominal wall just caudal to the costal arch at the level of the fundus of the stomach.
3. Pass the feeding tube half-way through the incision so that the mushroom tip is within the abdomen and the catheter tip outside.
4. Place a full-thickness purse-string suture in the fundus, midway between the lesser and greater curvatures. The diameter of the purse string should be large enough to facilitate passage of the feeding tube through the centre. Do not tie the ends of the suture, but hold them with haemostats.
5. Make a stab incision in the centre of the purse-string suture, just large enough to push the folded mushroom tip of the catheter through.
6. Tighten the purse-string suture and secure it around the shaft of the gastrostomy tube; this seals the stomach wall around the tube.
7. Place four horizontal mattress sutures around the feeding tube between the stomach wall and the transversus abdominis muscle in a square configuration, to appose the stomach to the left abdominal wall. Place all the sutures before any is tied.
8. Omentum can be wrapped around the gastropexy site to promote healing.
9. Pull the external (catheter tip) end of the tube so that the mushroom tip is positioned snugly against the stomach wall.
10. Secure the tube to the skin using a Chinese finger-trap suture.
11. Close the coeliotomy wound routinely.

**Tube care and maintenance; Feeding technique; Tube removal; Potential complications**

As for **Gastrostomy tube placement – (a) endoscopic**

Further details on placement and use of gastrostomy tubes can be found in the *BSAVA Manual of Canine and Feline Gastroenterology* and the *BSAVA Manual of Canine and Feline Rehabilitation, Supportive and Palliative Care.*
Haemagglutination test

**Indications/Use**
- Confirmation of suspected immune-mediated haemolytic anaemia

**Equipment**
- 1 ml of venous blood, freshly collected in an EDTA tube (see Blood sampling – (a) venous)
- Microhaematocrit tube
- Microscope slide and coverslip
- 0.9% saline
- 2 ml syringe and needle
- Microscope

**Technique**
1. Using a microhaematocrit tube, place 2–4 drops of blood on a microscope slide.
2. Using a syringe and needle, withdraw saline from a bag and add 2–4 drops to the blood on the slide.
3. Rock the slide gently for 1 minute.
4. Examine the slide grossly for agglutination, i.e. clumping of red cells.
5. Add a coverslip and examine microscopically under high power.
6. A positive result is the microscopic visualization of clumps of randomly oriented red blood cells.

It is very important to examine the slide using the microscope, as rouleaux formation appears grossly identical to true agglutination.

Further details of the test and its interpretation are given in the **BSAVA Manual of Canine and Feline Clinical Pathology**.

**High-dose dexamethasone test** see
- Dexamethasone suppression test – (b) high dose
Hip luxation – closed reduction

Indications/Use
• Recent traumatic hip luxation

Contraindications
• If luxation has been present for longer than 7–10 days, closed reduction is unlikely to be successful

Equipment
• Soft cotton rope or towel sling
• Sandbag
• As for Ehmer sling

Patient preparation and positioning
• General anaesthesia is required.
• Ventrodorsal and lateral radiographs of the pelvis should be obtained to confirm hip luxation and to evaluate the direction of the luxation. Radiography is also required to detect avulsion fractures of the teres ligament, other pelvic fractures and the presence of pre-existing coxofemoral osteoarthritis, which may complicate management of hip luxation.
• The patient should be positioned in lateral recumbency, with the affected limb uppermost.
• For large dogs it may be beneficial to secure their hindquarters to the table, using a soft cotton rope placed within the inguinal region of the affected limb and secured caudodorsally to the table. Alternatively, the affected pelvic limb may be supported using a towel sling: the towel is passed through the inguinal region and both ends are held by an assistant standing on the dorsal side of the patient.

Technique
Craniodorsal luxation (the majority of cases)
1. Manipulate the femoral head to loosen any adhesions and apply caudoventral traction to the stifle region to counteract muscle contraction.
2. Extend the leg, adduct and externally rotate it with continued traction to lift the femoral head over the dorsal rim of the acetabulum and reduce the hip. The greater trochanter may be guided caudally and distally with the non-dominant hand.
3. A palpable/audible click may be noted at reduction. Apply pressure to the trochanter and rotate the joint to express any haematoma from the joint space.
4. Manipulate the hip and check it for range of motion and stability.
5. Confirm reduction radiographically.
6. If appropriate, apply an Ehmer sling for 7–10 days.
Cranioventral luxation
- Manipulate the femoral head to the craniodorsal position and reduce as described above.
- *Alternatively,* it may be possible to manipulate the femoral head directly back into the acetabulum by palpation.
- Confirm reduction radiographically.

Caudoventral luxation
- In this instance the femoral head often sits within the obturator foramen. Traction is applied to the affected limb by grasping it proximal to the stifle whilst counter-traction is applied to the tuber ischium. This releases the femoral head from the obturator foramen.
- The femoral head is then lifted laterally and cranially into the acetabulum. This may be aided by placing a sandbag between the thighs: the sandbag is then used as a fulcrum to lever the femoral head back into the acetabulum.
- Confirm reduction radiographically.
- An Ehmer sling should *not* be used in these cases.

Aftercare
- Exercise should be restricted for 3–6 weeks following closed reduction of hip luxation, to allow for healing of the periarticular tissues.
- Appropriate analgesia should be provided.

Potential complications
- Reluxation of the hip
- Hip osteoarthritis
- Iatrogenic damage to the cartilage of the femoral head

Further information on hip problems and their treatment can be found in the *BSAVA Manual of Canine and Feline Musculoskeletal Disorders.*
Intraosseous cannula placement

Indications/Use

- Fluid administration:
  - Where direct intravenous access is not possible (e.g. hypovolaemic puppies/kittens, severe vascular collapse)
  - To provide *initial* fluid resuscitation and medication until intravenous access is possible

- Most substances that can be given intravenously can be given into the medullary space, and absorption into the vasculature is extremely rapid.
- However, hypertonic and alkaline fluids may cause pain and lameness.

Contraindications

- Sites with high risk of bacterial contamination/infection from e.g. local tissue damage, local skin infection, diarrhoea, urinary incontinence
- Where intravenous access is possible

Equipment

- As required for *Aseptic preparation – (a) non-surgical procedures*
- Potential intraosseous cannulas:
  - Commercially available intraosseous cannulas
  - Spinal needle
  - Bone marrow aspiration needle
  - Hypodermic needle (may be able to penetrate the soft cortex of young puppies and kittens)

Ideally, intraosseous cannulas should have a central stylet to prevent a core of bone from obstructing the cannula

Cannula size should be appropriate for the estimated diameter of the bone marrow canal, size of the point of access into the bone and proposed fluid administration rates

- No. 11 scalpel
- Local anaesthetic
- T-connector or extension set containing heparinized saline (1 IU of heparin per ml of 0.9% saline)
- Sterile non-adherent dressing or swab
- Adhesive tape or suture
- Soft padded bandage and outer protective bandage

Patient preparation and positioning

- The animal’s limb must be held still; this can usually be achieved by manual restraint but sedation may be required.
Aseptic preparation – (a) non-surgical procedures should be carried out on a large area of skin surrounding the proposed site of cannula entry.

Possible sites:
- The medial aspect of the trochanteric fossa of the femur
- The flat medial surface of the proximal tibia, 1–2 cm distal to the tibial tuberosity
- The cranial aspect of the greater tubercle of the humerus (see Bone marrow aspiration)
- The wing of the ilium (see Bone marrow aspiration).

The preferred sites are those in the femur and tibia.

**Technique**
1. Infiltrate local anaesthetic down to the level of the periosteum.
2. Make a stab incision in the skin over the proposed entry point into the bone.
3. Insert the cannula through the skin and directly down on to the bone.
4. Insert the cannula into the bone, using a firm twisting motion until the cannula has passed through the cortex and a small way into the medullary cavity. Entry into the medullary cavity is detected as a decrease in resistance to cannula insertion into the bone or increased stability of the cannula within the bone. When the cannula is properly seated within the medullary cavity, movement of the cannula will result in the same movement of the bone.
5. Remove the stylet, if present.
6. Flush the cannula with heparinized saline, and attach a T-connector or infusion set.
7. Secure the cannula to the surrounding skin with sutures or tape.
8. Cover the entry site through the skin with a sterile swab or sterile non-adherent dressing.
9. Apply a bandage to the limb to protect the cannula.

Care of intraosseous cannulas
- Intraosseous cannulas are most often a short-term solution.
- Catheter patency should be maintained by any fluid running through it. If the catheter is not being used continuously, intermittent flushing with saline or heparinized saline should be performed several times a day (up to every 4 hours), as well as before and after use. If a catheter becomes blocked it should be removed.
- The bandage should be replaced and the cannula examined at least twice a day, using aseptic technique.
- The site of insertion should be monitored for signs of heat, erythema, swelling, pain or subcutaneous leakage of fluid. If these signs are noted the cannula should be removed.
- The cannula should be removed as soon as it is no longer required.

Potential complications
- Sciatic nerve damage: this can be avoided during placement of a femoral cannula, by walking the needle off the medial edge of the greater trochanter
- Growth plate damage in young animals
- Cannula displacement
- Subcutaneous leakage of fluids or medications
- Infection

Intravenous catheter placement – (a) peripheral veins

Indications/Use
- Intravenous fluid administration
- Intravenous drug administration
- Repeat intravenous blood sampling, although a central line is often preferable for this indication
Contraindications
- Sites with a high risk of bacterial contamination/infection, e.g. due to local tissue damage, local skin infection, diarrhoea, urinary incontinence

Equipment
- Over-the-needle intravenous catheter, comprising a needle (or stylet) with a closely associated catheter fitted over the needle. Fluid flow rate is related to catheter length and radius. For rapid fluid administration, choose the shortest catheter with the largest radius that can pass into the vein:
  - Dogs: usually 22 G (blue), 20 G (pink) or 18 G (green)
  - Cats: usually 22 G (blue)
  - Puppies/Kittens or patients with collapsed veins may require 24 G (yellow)
- No. 11 scalpel
- T-connector or extension set containing heparinized saline (1 IU of heparin per ml of 0.9% saline) or injection cap
- Adhesive tape
- Soft padded bandage and outer protective bandage
- 4% chlorhexidine gluconate or 10% povidone–iodine
- 70% surgical spirit

Patient preparation and positioning
- An area of skin surrounding the point of vein entry should be clipped. Long hair on the caudal aspect of the limb may need to be removed if it will interfere with securing the catheter, and to help prevent contamination. In some dogs a complete 360-degree clip of the limb may be necessary.
- Using cotton wool or gauze swabs, the skin over the vein is cleaned with chlorhexidine or povidone–iodine and then sprayed with surgical spirit.
- Possible sites:
  - Cephalic vein and accessory cephalic vein in the distal forelimb (see Blood sampling)
  - Medial and lateral saphenous veins in the distal hindlimb (see Blood sampling)
  - Auricular veins in breeds with large ears, e.g. Basset Hound
  - Dorsal common digital vein of the metatarsal bones.
- The animal’s limb must be held still. This can usually be achieved by manual restraint, but sedation may be useful in animals whose temperament does not permit restraint.

Technique
Hands should be washed with an antiseptic solution prior to catheter placement. Sterile gloves are not necessary, but the precise point of catheter insertion should not be contaminated after aseptic preparation.
1. The vein is raised by an assistant by placing pressure over the vein proximal to the site of catheter placement.
2. A small cut can be made in the skin over the vein but is not usually required except in dehydrated animals or those with very thick skin.
3. Advance the over-the-needle catheter through the skin into the vein, at an angle of 30–40 degrees, with the bevel of the stylet uppermost.
4. Once blood is visualized in the flash chamber of the catheter, the stylet and catheter are flattened (i.e. the angle between the catheter and the limb is reduced). Then advance the stylet/catheter unit a small distance further into the vein to ensure that the catheter lies fully within the lumen.
5. Hold the stylet in position while advancing the catheter forward off the stylet and completely into the vein.
6. Remove the stylet.
7. The assistant can now occlude the vein by applying pressure over it at the distal end of the catheter to prevent spillage of blood.
8. Connect a T-connector or extension set containing heparinized saline or an injection cap to the catheter to close it.
9. Secure the catheter firmly in place with adhesive tape, and cover with a bandage.

Care of peripheral venous catheters
- The bandage should be replaced and the catheter examined at least twice a day.
- The site of insertion should be monitored for signs of heat, erythema, swelling, pain or leakage of fluid. If signs of phlebitis are present the catheter should be removed.
- The leg and foot should be checked for swelling above the catheter site (indicating extravasation of fluid) and swelling of the toes (indicating the bandage or tape is too tight). Either of these complications necessitates catheter removal.
- Catheter patency should be maintained by any fluid running through it. If the catheter is not being used continuously, intermittent flushing with heparinized saline should be performed several times a day (up to every 4 hours) as well as before and after use. If a catheter becomes blocked it should be removed.
- When a catheter is not in use, a sterile injection cap should be used to close the catheter from the environment. Disconnection of fluid lines should be avoided and only performed when absolutely necessary to reduce contamination of the catheter.

Potential complications
- Catheter displacement/extravasation of fluids or medications
- Phlebitis/thrombophlebitis
- Thrombosis/thromboembolism
- Infection
- Dislodgement of catheter
- Air embolism
- Exsanguination
Intravenous catheter placement – (b) jugular vein (modified Seldinger technique)

Indications
- Long-term (> 5 days) administration of intravenous fluids
- Administration of intravenous hypertonic fluids or medications
- Repeated blood sampling
- For central venous pressure measurements
- Where maintenance of a peripheral catheter may be challenging (e.g. due to patient conformation or temperament)

Contraindications
- Coagulopathy
- Local tissue damage or skin infection in the ventral cervical region

Equipment
- As required for Aseptic preparation – (a) non-surgical procedures
- Sterile gloves
- Central venous Seldinger catheter pack
- Over-the-needle catheter of appropriate diameter to receive the guide wire, if preferred to the introducer needle in the Seldinger pack
- No. 11 scalpel
- Heparinized saline (1 IU of heparin per ml of 0.9% saline) in two or three 5 ml syringes
- Suture materials
- Sterile dressing
- Bandaging material
- ECG monitoring equipment

Patient preparation and positioning
- Although central lines may be placed in conscious patients if they are weak or debilitated, sedation or anaesthesia is required in most animals to prevent movement during the procedure.
- Anaesthetized animals should be placed in lateral recumbency, with a sandbag positioned under the neck and the jugular vein to be catheterized uppermost.
Aseptic preparation – *(a) non-surgical procedures* is necessary, with the use of a fenestrated drape.

**Technique**

1. With their hand under the sterile drape, an assistant raises the jugular vein.

2. Make a stab incision over the jugular vein.

3. Insert the introducer needle or an over-the-needle catheter into the vein in a rostrocaudal direction. If using an over-the-needle catheter, then remove the needle. The assistant should then stop raising the jugular vein.

4. Insert the guide wire through the introducer needle or catheter into the jugular vein.
   - The guide wire is usually held within an adapter to aid placement and may have a J-shaped tip. The J-shaped tip should be straightened by withdrawal into the adapter prior to insertion into the introducer needle or catheter.
   - The ECG should be monitored for arrhythmias, as overlong wires can enter the heart.
– Care must be taken to ensure that the guide wire is held at all times for the remainder of the procedure to prevent loss of the wire into the vein.

5. Remove the introducer needle or catheter, leaving the guide wire in place.

6. Advance the vessel dilator over the guide wire and into the vein to enlarge the subcutaneous tunnel; then remove the dilator.

7. Advance the catheter over the wire and into the vein.

8. Remove the guide wire.

9. Place sterile injection caps, ideally incorporating on/off valves, on the end of each port of the catheter. Withdraw any air, plus some blood, from each port into a syringe part-filled with heparinized saline, to prevent air embolism and ensure intravascular placement. Then, immediately flush through each port with heparinized saline.

10. Secure the catheter to the patient with sutures placed through specific suture grooves or holes in a suture wing at the base of the catheter.
11. Place a sterile dressing over the entry site of the catheter, and bandage the catheter carefully in place. The bandage should not be too tight; it should be possible to pass a hand under it comfortably.

**Care of jugular catheters**

- The bandage should be replaced and the catheter examined at least once a day. The site of insertion should be monitored for signs of heat, erythema, swelling, pain or leakage of fluid. If signs of phlebitis are present, or the animal develops unexplained pyrexia, the catheter should be removed and the tip sent for microbiological culture.
- Catheter patency should be maintained by any fluid running through it. If the catheter is not being used continuously, intermittent flushing with saline or heparinized saline (1 IU of heparin per ml of 0.9% saline) should be performed several times a day (up to every 4 hours) as well as before and after use.
- When a catheter is not in use, sterile injection caps should be used to close access ports; **ports should never be left open to the air**. Disconnection of fluid lines should be avoided and only performed when absolutely necessary to reduce contamination of the catheter.

**Potential complications**

- Catheter displacement/extravasation of fluids or medications
- Phlebitis/thrombophlebitis
- Thrombosis/thromboembolism
- Infection
- Dislodgement of catheter
- Air embolism
- Exsanguination

**Intravenous urography**

**Indications**

- Evaluation of renal size, shape, position and internal architecture
- Demonstrating ureter position and patency
- Investigation of urinary incontinence
- Investigation of haematuria or pyuria NOT arising from the lower urinary tract

**Contraindications**

- Renal failure

**Equipment**

- As required for **Urethral catheterization** and **Intravenous catheter placement – (a) peripheral veins**
- Water-soluble contrast medium for intravenous use (see below for concentrations and dose rates)
- 20–50 ml syringe
• Giving set
• 500 ml bag of 0.9% saline

Patient preparation and positioning
• In an elective examination, withhold food for 24 hours before the procedure.
• Give at least one non-irritant cleansing enema (e.g. warm water) 2–3 hours prior to examination.
• Use general anaesthesia or heavy sedation, since injection of contrast medium in the conscious patient may cause retching, vomiting and struggling.
• Pass a urinary catheter and drain the bladder of urine (see Urethral catheterization). The catheter can be left in situ but should be pulled out of the bladder and into the proximal urethra.
• Positioning will be determined by views required (see below).

Technique
There are two basic techniques:
• High-concentration, low-volume (bolus) IVU, with or without abdominal compression: preferred for imaging the kidneys
• Low-concentration, high-volume (infusion) IVU: preferred for imaging the ureters. This technique may produce inferior renal opacification but gives improved visibility of the ureters due to increased osmotic diuresis.

High-concentration, low-volume (bolus) IVU:
1. Obtain lateral and ventrodorsal plain radiographs of the abdomen to check exposure factors and to allow comparison with subsequent contrast images.
2. Position the patient in dorsal recumbency, as the ventrodorsal view is more helpful initially.
3. Inject contrast medium at room temperature or warmed to 37°C (to reduce viscosity) at 300–400 mg iodine/ml (concentration is given as a number after the trade name) rapidly into a cephalic vein via a catheter. Dose rate is 850 mg iodine/kg bodyweight (i.e. about 2 ml/kg).
4. Immediately take the first radiograph.
5. Take further ventrodorsal radiographs at regular intervals (e.g. 2, 5, 10 and 15 minutes) until a clear nephrogram and pyelogram are obtained. Then obtain a lateral view ± oblique views to visualize the ureters.
6. Continue until a diagnosis is made or a normal appearance is confirmed.

Low-concentration, high-volume (infusion) IVU:
1. Obtain lateral and ventrodorsal plain radiographs to check exposure factors and to allow comparison with subsequent contrast images. If the study is to demonstrate the location of the ureter endings in incontinent patients, a pneumocystogram should be obtained to outline the bladder neck.
2. Administer contrast medium at room temperature or warmed to 37°C (to reduce viscosity) at 150 mg iodine/ml (more concentrated media can be diluted with 0.9% saline) over 5–30 minutes into the cephalic vein via a catheter. Dose rate is 1200 mg iodine/kg bodyweight (i.e. 8 ml/kg).

3. Obtain radiographs as above, starting approximately 5 minutes into the infusion until a diagnosis is made, or a normal appearance is confirmed.

**Potential complications**

- Anaphylaxis (rare): risk proportional to speed of injection, concentration of agent and volume infused
- Renal failure (rare): more likely with pre-existing renal disease; therefore ensure adequate hydration before and during the procedure

Further details of these procedures and their interpretation can be found in the *BSAVA Manual of Canine and Feline Abdominal Imaging* and the *BSAVA Manual of Canine and Feline Nephrology and Urology*.

**Iodinated contrast media**

Iodinated contrast media appear radiopaque on a radiograph due to iodine having a higher atomic number than soft tissues. There is a wide range of iodinated contrast media available. None is authorized for veterinary use and most are POM.

<table>
<thead>
<tr>
<th>Constituent</th>
<th>Properties</th>
<th>Trade name</th>
<th>Uses</th>
<th>Formulations (mg iodine/ml)</th>
</tr>
</thead>
<tbody>
<tr>
<td>Iotalamic acid</td>
<td>Monomer Ionic</td>
<td>Conray</td>
<td>Vascular, urinary</td>
<td>141; 202; 280; 400</td>
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<tr>
<td>Sodim meglumine diatrizoate</td>
<td>Monomer Ionic</td>
<td>Urografin</td>
<td>Vascular, urinary</td>
<td>146; 325; 370</td>
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<tr>
<td>Iohexol</td>
<td>Monomer Non-ionic</td>
<td>Omnipaque</td>
<td>Vascular, urinary</td>
<td>140; 180; 240; 300; 350</td>
</tr>
</tbody>
</table>

**Use**

- Contrast studies of the GI tract and urinary tract, angiography, arthrography, sialography, dacryocystography, sino- or fistulography and myelography
• Only non-ionic agents may be used for myelography, but not all non-ionic agents are authorized for myelography in humans
• Fluid and electrolyte disturbances should be corrected prior to use

Contraindications
• Known or suspected sensitivity to iodine-containing preparations
• Uncontrolled hyperthyroidism is a contraindication in humans
• Should be used with care in animals with moderate to severe impairment of renal function. Primarily excreted via kidneys and pre-existing renal disease predisposes to contrast media-induced renal failure

Adverse reactions
Anaphylaxis may occur following i.v. injection of any iodinated contrast medium. Nausea, vomiting, hypotension, dyspnoea, erythema, urticaria, sensation of heat, and cardiac rate or rhythm disturbances have been reported in humans following i.v. injection. Seizures or transient motor or sensory dysfunction have been reported in humans following myelography. Aspiration of high osmolar contrast media may result in pulmonary oedema. May aggravate clinical signs in patients with myasthenia gravis. These effects are more common with ionic than with non-ionic media and with hyper-osmolar media. Non-ionic low-osmolar and iso-osmolar agents have fewer adverse effects on the cardiovascular system and are safer in neonates and animals with cardiovascular disease. When used intravascularly, catheters should be flushed regularly to minimize risk of clotting; non-ionic media have less anticoagulant activity in vitro than ionic preparations. Extravasation of high-osmolar agents may rarely lead to soft tissue injury. Warming the contrast media prior to injection reduces minor side effects. Specific formulations for GI contrast studies cannot be used for other examinations due to the presence of additives.

Drug interactions
Iodinated contrast media decrease thyroid uptake of iodine and preclude therapeutic radioiodine therapy for 2 months following administration. Hypersensitivity reactions may be aggravated in patients on beta blockers.

Iodine contrast studies see
• Intravenous urography
• Retrograde urethrography/vaginourethrography

IV see
• Intravenous
Joint tap see
- Arthrocentesis
Myringotomy

**Indications**
- To obtain samples for diagnostic evaluation of otitis media
- To permit lavage of the tympanic bulla for non-surgical management of otitis media

**Contraindications**
- Any animal with a normal appearance to the tympanic membrane and no evidence of otitis media

**Equipment**
- Otoscope with a sterilized otoscopic speculum; a video-otoscope is best but not essential
- Sterile catheters (e.g. 3.5 Fr tom cat catheter, long polypropylene catheter, 3.5 Fr feeding tube) of sufficient length to access the tympanic membrane via the working channel of a video-otoscope or along the speculum of an ordinary otoscope
- 2.5 or 5 ml, plus two 20 ml syringes
- 3-way tap
- Sterile saline
- Otic ceruminolytic agent (may be needed in dogs)
- Bacteriological swab
- EDTA sample pots
- Microscope slides

**Patient preparation and positioning**
- In chronic otitis externa, a short course of systemic glucocorticoids may aid optimal visualization of the tympanic membrane.
- General anaesthesia is required.
- A properly placed and inflated endotracheal tube is recommended due to possible drainage of fluid from the middle ear canal into the pharynx.
- The animal is placed in lateral or sternal recumbency. A towel may be used to elevate the caudal head and neck slightly in relation to the muzzle.
- Protect the animal's eyes if ceruminolytics are used to clean the external ear canal.

**Technique**
Personnel should wear gloves, face mask and, ideally, eye protectors to avoid contact with contaminated aerosols.
1. Clean and dry the external ear canal.
   - Cleaning can be performed safely with sterile saline.
   - Ceruminolytic agents can be used in dogs to speed removal of waxy exudates but must be flushed completely from the ear canal with sterile saline.
   - Ceruminolytics should not be used to flush the ears of cats if the integrity of the tympanic membrane is uncertain, due to the higher risk of neurological complications.
   - Drying can be achieved by suction of lavage solutions.
2. Insert the otoscope into the external ear canal to visualize the tympanic membrane. This requires straightening of the external ear canal, by pulling the pinna dorsally and outwards away from the head.
3. Under direct visualization, flush the ear canal with sterile saline to remove any remaining debris.
   - A long polypropylene catheter attached to a 3-way tap and two 20 ml syringes can achieve efficient flushing and suction.
   - The person controlling the otoscope visualizes and positions the tip of the catheter, while an assistant flushes in saline with one syringe and removes the fluid by suction through the second syringe.
4. The site of myringotomy should be ventral at the 6 o’clock to 7 o’clock position. Position the otoscope carefully to visualize this site.
5. Cut the tip of the catheter at an angle to make it sharp enough to incise the tympanic membrane.
6. Under direct visualization, pass the tip of the catheter through the ventral aspect of the tympanic membrane into the middle ear and maintain it in this position.
7. An assistant should infuse 1 ml of sterile saline via the catheter into the middle ear and then aspirate to obtain a fluid sample for cytology and culture.
8. The assistant should then flush the middle ear gently with sterile saline until the fluid aspirated back is clear.
9. Remove the catheter from the middle ear.
10. Remove the otoscope.

**Potential complications**
- Neurological signs (including head shaking, Horner’s syndrome, deafness) can result from over-aggressive lavage or the use of ceruminolytics.

This technique is illustrated in the *BSAVA Manual of Canine and Feline Endoscopy and Endosurgery* and cytological findings are discussed in the *BSAVA Manual of Canine and Feline Clinical Pathology*.
Nasal oxygen administration

Indications/Use
- Patients requiring oxygen for several days
- Patients that do not tolerate an oxygen mask
- Patients that are too large to fit inside an oxygen cage

Contraindications
- Inspired oxygen concentrations may not be high enough for very hypoxic animals, particularly if they are mouth-breathing. In very hypoxic animals, bilateral nasal oxygen lines can be used
- Not useful for brachycephalic animals or patients with facial disease or pain

Equipment
- Rubber catheter/soft polythene feeding tube: large dogs: 5–10 Fr, depending on the size of the animal; 5 Fr in cats and dogs <5 kg
- Nasal prongs
- Topical local anaesthetic
- Sterile aqueous lubricant (e.g. K-Y jelly)
- Non-absorbable suture material, needle and needle-holders
- Tissue glue
- 25 mm wide adhesive tape
- Oxygen delivery system
- Humidifier (container filled with distilled water)
- Elizabethan collar

Patient preparation and positioning
- Usually performed in the conscious animal, although fractious animals may benefit from light sedation.
- The patient is positioned in sternal recumbency, or sitting.
- Apply several drops of local anaesthetic down one nostril and wait approximately 10 minutes before inserting the catheter.

Technique

Nasal catheter
1. Measure the distance from the nares to the medial canthus of the eye and mark this on the catheter.
2. Following desensitization of the nostril with a topical anaesthetic, gently insert the lubricated catheter into the nostril in a ventromedial direction (toward the base of the opposite ear) and advance it to the mark. The nasal planum can be pushed dorsally to direct the tube ventrally.
3. Once the catheter is in place, it is contoured around the alar fold, and sutured or glued in place on the side of the face. For the most secure placement, a suture should be placed as close to the nasal-cutaneous junction as possible.

4. Attach the nasal catheter to an oxygen delivery system with flow rates of 100–200 ml/kg/minute.

5. Humidify the oxygen by bubbling it through a chamber filled with distilled water.

6. Place an Elizabethan collar.

**Nasal prongs**

- Some animals can be best managed using human bilateral nasal ‘prongs’ that penetrate 1 cm or less into the nasal cavity.
- Inspired oxygen concentrations of 30–50% can easily be achieved using this type of system, although panting probably limits the effectiveness of prongs.

**Potential complications**

- Long-term therapy with high concentrations of oxygen ($F_{O_2} > 0.6$ for >12 hours, or sooner with assisted ventilation) can be associated with lung damage (‘oxygen toxicity’). Although rare, every effort should be made to minimize $F_{O_2}$ for critically ill patients
- Sneezing and dislodgement of catheter or prongs
- Nasal discharge is common but is not clinically significant

Further information on administration of supplementary oxygen, including the use of masks and oxygen cages, can be found in the *BSAVA Manual of Canine and Feline Anaesthesia and Analgesia* and the *BSAVA Manual of Canine and Feline Emergency and Critical Care*.

**Naso-oesophageal tube placement**

**Indications/Use**

- Short-term nutritional support (2–3 days) of cats or small dogs. In large dogs, the sheer volume of a liquid diet required to meet the energy needs is frequently limiting
- Animals that require assisted feeding but in which general anaesthesia is contraindicated

**Contraindications**

- Comatose, recumbent or dysphoric animals at risk of aspiration
- Animals with facial trauma
- Animals with persistent vomiting
• Severe nasal disease or dysphagia
• Oesophagitis or severe oesophageal dysfunction (e.g. megaoesophagus)

Equipment
• Naso-oesophageal tube
  – Cats: 3.5–5 Fr
  – Dogs: 3.5–8 Fr
• 5–10 ml syringe
• Topical local anaesthetic gel and drops
• 25 mm wide adhesive tape
• Suture materials
• Elizabethan collar

Patient preparation and positioning
• The procedure should be performed with the animal conscious or under light sedation.
• The patient can be positioned standing, or in lateral or sternal recumbency.
• The animal’s head should be placed in a ‘neutral’ position (i.e. not too flexed or extended).

Technique
1. Instil anaesthetic drops into one nostril.
2. Measure the distance from the nose to the 7th intercostal space and mark this on the tube with a piece of tape.
3. Lubricate the tube with anaesthetic gel to aid insertion.
4. Aim the tube ventromedially (toward the base of the opposite ear) so that it will pass into the ventral meatus of the nasal cavity. The nasal planum can be pushed dorsally to direct the tube ventrally.
5. Pass the tube until it reaches the predetermined position (nose to 7th intercostal space). It is very important to check its position carefully. If the tube is in the oesophagus, there should be negative pressure when suction is placed on the tube using a 5–10 ml syringe. As an additional test, instil a small amount of water into the tube before administering food.
6. A lateral radiograph can be taken to ensure not only that the tube is in the proper location (and ends in the distal third of the oesophagus, not the stomach), but also that the tube is not kinked or twisted.
7. Make two butterfly tape strips using 25 mm wide adhesive tape and attach these to the tube.
8. Suture or tissue glue these tape strips to the dorsal aspect of the muzzle and the top of the head.
9. Place an Elizabethan collar.

Feeding
• Feeding can commence immediately after tube placement.
• Before feeding each time, the tube should be flushed with small amounts (5–10 ml) of lukewarm tap water. If the animal coughs,
feeding should not be continued as the tube may have become dislodged. Radiographs should be taken to confirm the tube position.

- As the feeding tubes are of narrow bore, a liquid or semi-liquid food must be used.
- Food should be warmed to body temperature and injected into the tube over several minutes.
- Feed only half of the calculated daily calorific requirement (see Resting energy requirement) on the first day.
- On the second day, increase feeding to the calculated calorific intake, if tolerated.
- Divide the daily requirement into multiple (5 or 6) feeds per 24 hours.
- Flush the tube after feeding with 5–10 ml of lukewarm tap water.

Maintenance and removal of feeding tubes

- Twice a day, the external nares should be wiped gently with cotton wool soaked in water.
- Carbonated drinks can be used to clear blocked tubes.
- The tube can be removed at any time following placement: sutures holding the tube should be cut and the tube removed in one motion.

Potential complications

- Rhinitis
- Vomiting
- Regurgitation
- Aspiration of oesophageal contents
- Tube dislodgement

Further information on assisted feeding can be found in the BSAVA Manual of Canine and Feline Gastroenterology and the BSAVA Manual of Canine and Feline Rehabilitation, Supportive and Palliative Care.

Neurological examination

Indications/Use

- To diagnose and localize disorders of the nervous system
- To provide information on the severity of any disorder

Equipment

- Reflex hammer
- A bright light source
- Artery forceps
- Hypodermic needle
- 70% surgical spirit
Patient preparation and positioning

• The neurological examination should be performed with the animal conscious.
• Animals should be kept calm throughout the examination.
• Positioning will vary, depending on the part of the examination being performed.

Observation

The following parameters should be observed before starting the ‘hands-on’ part of the examination:

• Mental status and behaviour
• Posture and body position at rest
• Gait (see Orthopaedic examination)
• Abnormal involuntary movements.

Cranial nerve assessment

Olfactory nerve – CN I
Smell is assessed by testing the animal’s response (sniffing or licking of the nose, aversion of the head) to aromatic substances, e.g. surgical spirit whilst blindfolded. Care should be taken not to use irritating substances that could stimulate the trigeminal nerve and cause similar responses.

Optic nerve – CN II
Vision tests are described in Ophthalmic examination.

Oculomotor nerve – CN III
• Observe eye position and movements (a) at rest and (b) by testing for normal physiological nystagmus (vestibulo-ocular reflex) by moving the head from side to side as well as up and down (see Vestibulocochlear nerve – CN VIII). Normal physiological nystagmus (also called ‘jerk’ nystagmus) is an involuntary rhythmic movement of the eyes, which typically presents with a slow phase in one direction and a quick phase in the other direction.
• The parasympathetic function of CN III can be assessed by observation of the pupil size and evaluation of the pupillary light reflex (PLR; see Ophthalmic examination). This tests the integrity of the optic nerve to the level of the lateral geniculate nucleus and should be assessed in all blind animals to determine lesion location.

Trochlear nerve – CN IV
Observe the eye position at rest and test for normal physiological nystagmus (see above). Lesions of the trochlear nerve produce a dorsolateral strabismus (extorsion) of the contralateral eye.

Trigeminal nerve – CN V
• The motor function of CN V is assessed by evaluating the size and symmetry of the masticatory muscles and testing the resistance of the jaw to opening the mouth.
• The sensory function (sensation of the face) can be individually tested by:
  – The corneal reflex observed on touching the cornea (ophthalmic branch)
  – The palpebral reflex observed on touching the medial or lateral canthus of the eye (ophthalmic or maxillary branch, respectively)
  – The response to stimulation of the nasal mucosa (ophthalmic branch).

A reflex response is best distinguished from a conscious response by stimulating the nasal mucosa: the normal animal will pull its head away, while the animal with forebrain disease may blink or show a facial twitch, but not show a conscious reaction.

**Abducent nerve – CN VI**
Observe the eye position and movement at rest; test for normal physiological nystagmus (see above); and test for normal eyeball retraction during the corneal reflex. Lesions of the abducent nerve result in: an ipsilateral convergent strabismus; an inability of the eye to cross the midline when evaluating the horizontal physiological nystagmus; and an inability to retract the eyeball.

**Facial nerve – CN VII**
• The motor function of CN VII is primarily assessed by observation of the face for symmetry (position of the ears and lip commissure on each side within the same plane, symmetry of the palpebral fissure), spontaneous blinking and movement of the nostrils. Motor dysfunction produces: ipsilateral drooping of and inability to move the ear and lip; widened palpebral fissure and absent spontaneous and provoked blinking; absent abduction of the nostril during inspiration; and deviation of the nose toward the normal side due to the unopposed muscle tone on the unaffected side.
• The Schirmer tear test can evaluate the parasympathetic supply of the lacrimal gland associated with CN VII. Examining the mouth for a moist mucosa can subjectively assess salivation.

**Vestibulocochlear nerve – CN VIII**
• Observe the animal’s body and head posture at rest, and evaluate its gait (see Orthopaedic examination). This function can also be more specifically assessed by testing the vestibulo-ocular reflex (see above). Vestibular dysfunction may result in any or all of the following: head tilt; falling; leaning; rolling; circling; abnormal and/or positional nystagmus; positional strabismus; and asymmetrical ataxia.
• The auditory function of CN VIII is difficult to assess. The startle reaction consists of observing the animal’s response to noise (e.g. handclap, whistle), but this does not detect unilateral or partial deafness. The best assessment is the animal’s response to noise when asleep: most owners can report whether they have to touch their animal in order to wake them. Electrophysiological assessment is necessary to confirm and assess the severity of the hearing loss.
Glossopharyngeal and vagus nerves – CN IX and CN X
- The *pharyngeal* (swallowing or gag) reflex can be evaluated by:
  - Applying external pressure to the hyoid bones to stimulate swallowing
  - Stimulating the pharynx with a finger to elicit a gag reflex
  - Watching the animal eat or drink
  - Opening the animal’s mouth wide.
- CN IX dysfunction results in dysphagia, absent gag reflex and reduced pharyngeal tone.
- CN X dysfunction results in dysphagia, inspiratory dyspnoea (due to laryngeal paralysis), voice changes (dysphonia) and regurgitation (due to megaoesophagus in the case of bilateral vagal disorder).
- The *parasympathetic* portion of CN X can be evaluated by testing the oculocardiac reflex. This is achieved by applying digital pressure to both eyeballs and observing simultaneously a reflex bradycardia (also mediated by CN V).

Accessory nerve – CN XI
Lesions of this nerve result in atrophy of the trapezius muscle. Observe the position of the neck, as this may be deviated toward the affected side in chronic cases. Isolated lesions of the accessory nerve are an extremely rare finding.

Hypoglossal nerve – CN XII
- Inspect the tongue for atrophy, asymmetry or deviation to one side.
- Manually stretching the tongue and observing a voluntary retraction assesses the tongue’s tone.
- Applying food paste to the nose and observing the animal licking assesses the tongue’s movement.
- Lesions affecting CN XII can result in problems with prehension, mastication and deglutition.

Postural reaction testing
Proprioceptive positioning
- Place the paw in an abnormal position (turned over so that the dorsal surface is in contact with the ground) and determine how quickly the animal corrects it. The animal should be standing squarely on all four limbs, with the majority of its weight supported by an assistant.
- Place a piece of paper under a weight-bearing foot and pull it slowly in a lateral direction. A normal response is to pick up the limb and replace it in the correct position.

Hopping reaction
The hopping reaction is tested by holding the patient so that the majority of its weight is placed on one limb while the animal is moved laterally. Normal animals hop on the tested limb to accommodate a new body position as their centre of gravity is displaced laterally. An equal response should be seen on both sides.
Placing response

- Non-visual:
  i. Cover the animal’s eyes.
  ii. Lift the animal until the distal part of the thoracic limb is brought into contact with the edge of a table.
  iii. The normal response is for the animal immediately to place its foot on the table surface.

- Visual placing:
  i. Allow the animal to see the table surface.
  ii. Lift the animal toward the edge of the table.
  iii. Normal animals will reach for the surface before the paw touches the table.

Hemi-walking and wheelbarrowing

- Hemi-walking:
  - Tests the ability of the animal to walk on the thoracic and pelvic limbs of one side of the body, while you are holding the limbs on the other side
  - Push the animal away from the side on which its limbs are supported
  - Assess the speed and coordination of movements.

- Wheelbarrow:
  - Tests the thoracic limbs
  - Lift the pelvic limbs off the ground by supporting the animal under the abdomen and forcing it to walk forwards
  - Highlights subtle thoracic limb weakness and ataxia.

Spinal reflexes: muscle tone and size

Spinal reflex evaluation is performed to classify the neurological disorder as being of LMN or UMN type. This allows the examiner to localize the lesion to specific spinal cord segments or peripheral nerves. Spinal reflexes are best tested with the animal relaxed in lateral recumbency, with the side to be tested uppermost.

- **Lower motor neuron**: If LMNs are damaged, the following clinical signs are characteristically found:
  - Flaccid paresis and/or paralysis
  - Reduced or absent reflexes
  - Reduced or absent muscle tone
  - Early and severe muscle atrophy.

- **Upper motor neuron**: If UMNs are damaged, the following clinical signs are characteristically found:
  - Spastic paresis and/or paralysis
  - Normal to increased reflex activity (hyperreflexia)
  - Increased extensor muscle tone (hypertonia) manifested as a resistance to passive manipulation of the limbs
  - Chronic mild to moderate muscle atrophy (disuse atrophy).
Thoracic limbs

Withdrawal (flexor) reflex

This evaluates the integrity of spinal cord segments C6–T2 (and associated nerve roots) as well as the brachial plexus and peripheral nerves.

- The withdrawal reflex in the thoracic or pelvic limbs does not depend on the animal's conscious perception of noxious stimuli (nociceptive function).
- The withdrawal reflex is a segmental spinal cord reflex that only depends on the function of the local spinal cord segments.

- With the animal in lateral recumbency, apply a noxious stimulus by pinching the nailbed or digit with the fingers or a haemostat.
- This stimulus causes a reflex contraction of the flexor muscles and withdrawal of the tested limb.
- If this withdrawal reflex is absent, test individual toes to detect whether specific nerve deficits are present.
- When testing the flexor reflex, observe the contralateral limb for extension (crossed-extensor reflex), indicating a UMN lesion cranial to the C6 spinal cord segment.

Extensor carpi radialis reflex

This evaluates the integrity of spinal cord segments C7–T2 and associated nerve roots as well as the radial nerve.

- Strike the extensor carpi radialis muscle belly with a reflex hammer along the craniolateral aspect of the proximal antebrachium.
- The desired reaction is a slight extension of the carpus.

Biceps brachii and triceps reflexes

- Biceps reflex:
  - Place a finger over the distal end of the biceps brachii and brachialis muscle at the level of the elbow
  - Strike the finger with a reflex hammer
  - A normal reaction is flexion of the elbow or, at least, contraction of the biceps muscle.
- Triceps reflex:
  - Strike the triceps tendon proximal to its insertion on the olecranon
  - The desired reaction is an extension of the elbow or carpus.

Pelvic limbs

Withdrawal (flexor) reflex

This evaluates the integrity of spinal cord segments L4–S2 (and associated nerve roots) as well as the femoral and sciatic nerves.
• The withdrawal reflex in the thoracic or pelvic limbs does not depend on the animal’s conscious perception of noxious stimuli (nociceptive function).
• The withdrawal reflex is a segmental spinal cord reflex that only depends on the function of the local spinal cord segments.

• With the animal in lateral recumbency, apply a noxious stimulus by pinching the nailbed or digit with the fingers or a haemostat.
• This stimulus causes a reflex contraction of the flexor muscles and withdrawal of the tested limb.
• A normal reflex constitutes flexion of the hip (femoral nerve function), stifle and hock (sciatic nerve function).
• The hock must be extended in order to evaluate sciatic function (i.e. hock flexion).
• A crossed-extensor reflex in the pelvic limb indicates a UMN lesion cranial to the L4 spinal cord segment.

**Patellar reflex**
This evaluates the integrity of spinal cord segments L4–L6 (and associated nerve roots) as well as the femoral nerve.

• The animal is placed in lateral recumbency, with the stifle slightly flexed and the tested limb supported by placing one hand under the thigh.
• Striking the patellar tendon with a reflex hammer induces extension of the limb due to a reflex contraction of the quadriceps femoris muscle.
• A weak or absent reflex indicates a lesion of the L4–L6 spinal cord segments or the femoral nerve.

**Cranial tibial and gastrocnemius reflexes**
• The *cranial tibial reflex* is elicited by striking the proximal part of the cranial tibial muscle with a reflex hammer and observing for flexion of the tarsus.
• The *gastrocnemius reflex* is elicited by placing the finger over the gastrocnemius muscle and striking it with a hammer. The normal reaction is extension of the hock.

**Evaluation of the tail and anus**

**Perineal reflex**
Stimulation of the perineum with a haemostat should result in contraction of the anal sphincter, and flexion of the tail.

**Sensory evaluation**

**Cutaneous trunci (panniculus) reflex**
• Place the animal in sternal recumbency or in a standing position.
• Pinch the skin of the dorsal trunk between vertebral level T2 and L4–L5, and observe for a contraction of the cutaneous trunci muscles bilaterally, producing a twitch of the overlying skin.

• The reflex is present in the thoracolumbar region and absent in the neck and sacral region.
• Testing is begun at the level of the ilial wings: if the reflex is present at this level the entire pathway is intact and further testing is not necessary.
• With spinal cord lesions, this reflex is lost caudal to the spinal cord segment affected, indicating the presence of a transverse myelopathy. Pinching the skin cranial to the lesion results in a normal reflex, while stimulation of the skin caudal to the lesion does not elicit any reflex.

Deep pain perception
• Place the animal on its side, ideally with a second person talking to it or stroking it to distract its attention.
• Apply an initial gentle squeeze to the digits to elicit the withdrawal reflex.
• If the animal does not manifest any behavioural response following a gentle squeeze, apply heavy pressure to the bones of the digits with a haemostat.
• Only a severe bilateral spinal cord lesion impairs the sensation of deep pain. For this reason, testing of deep pain perception is a useful prognostic indicator in cases of spinal cord disease.
• This conscious pain perception must be assessed in all four limbs, the tail and the perineal region.
• The expected reaction is a behavioural response such as turning the head, trying to bite or vocalization.
• **Withdrawal of the limb is only the flexor reflex and should not be taken as evidence of pain sensation.**

More detail on these procedures and interpretation of the results can be found in the *BSAVA Manual of Canine and Feline Neurology.*
Oesophagostomy tube placement

Indications/Use
• Nutritional support for weeks to months

Contraindications
• Comatose, recumbent or dysphoric animals at risk of aspiration
• Persistent vomiting – the tube may be expelled or retroflexed into the nasopharynx
• Oesophagitis or severe oesophageal dysfunction (e.g. megaoesophagus)

Equipment
• As required for Aseptic preparation – (a) surgical procedures
• Oesophagostomy tube (red rubber tube, standard polyurethane feeding tube or silicone feeding tube):
  – Cats: 10–14 Fr; 23 cm long
  – Dogs: 14–24 Fr; 40 cm long
• OR Percutaneous oesophagostomy tube placement kit (containing oesophagostomy tube, rigid introducer and needle with peel-away sheath)
• Long curved forceps, e.g. Rochester–Carmalt
• No. 15 or 20 scalpel
• 25 mm wide adhesive tape
• Suture materials
• Sterile dressing to cover the tube site
• Light bandage for neck

Patient preparation and positioning
• General anaesthesia is required.
• The patient is placed in right lateral recumbency.
• Aseptic preparation – (a) surgical procedures from the dorsal to the ventral aspects of the neck is required over an area from the angle of the jaw to the shoulder.

Technique
Surgical cut-down

1. Insert curved forceps through the mouth and into the oesophagus, to the mid-cervical region.
2. Turn the tip of the forceps laterally and make a 5–10 mm skin incision over the point of the tips.

3. Bluntly dissect through the subcutaneous tissues and make an incision into the oesophagus over the tips of the forceps.

4. Push the tips of the forceps outwards through the incision to the external surface.

5. Measure the oesophagostomy tube from this point to the 7th intercostal space (distal oesophagus) and mark the tube with a piece of adhesive tape.

6. Open the tips of the forceps and grasp the distal end of the feeding tube.

7. Draw the end of the tube through the oesophagostomy incision and rostrally into the pharynx to exit the mouth.

8. Disengage the tips of the forceps, and curl the tip of the tube back into the mouth and feed it into the oesophagus.

9. Visually inspect the oropharynx to confirm that the tube is no longer present in the oropharynx.

10. The tube should slide easily back and forth a few millimetres, confirming that it has straightened.
12. Take a thoracic radiograph to confirm correct tube placement: the tip of the tube should be in the distal oesophagus, not the stomach.
13. Cover the tube site with a sterile dressing and place a soft, padded, loose, neck bandage.

**Using a percutaneous kit (van Noort technique)**

1. Insert the rigid introducer into the mid-cervical oesophagus via the oral cavity.
2. Rotate the introducer until you can feel the slot in its distal portion.
3. Make a small stab incision through the skin over the slot.
4. Introduce the needle with peel-away sheath through the skin incision into the distal portion of the rigid introducer.
5. Whilst holding the sheath in place, remove the needle from the sheath.
6. Remove the rigid introducer.
7. Measure the oesophagostomy tube from the needle entry point to the 7th intercostal space and mark the tube with a piece of adhesive tape.
8. Introduce the oesophagostomy tube through the sheath and down the oesophagus, stopping at the pre-marked length.
9. Grasping either side of the sheath, peel it away from the tube and remove it, to leave only the tube in place.
10. Secure the tube with a Chinese finger-trap suture.
11. Take a thoracic radiograph to confirm correct tube placement: the tip of the tube should be in the distal oesophagus, not the stomach.
12. Cover the tube site with a sterile dressing and place a soft, padded, loose, neck bandage.

**Feeding**

- **Feeding** can commence as soon as the patient has recovered from general anaesthesia.
- *Before feeding each time*, the tube should be flushed with small amounts (5–10 ml) of lukewarm tap water.
- As the feeding tubes are of narrow bore, a liquid or semi-liquid food must be used.
- The food should be warmed to body temperature and injected into the stomach over several minutes.
- Feed only *half* of the calculated daily calorific requirement (see **Resting energy requirement**) on the first day.
- On the second day, increase feeding to the calculated calorific intake, if tolerated.
- Divide the daily requirement into multiple (5 or 6) feeds per 24 hours.
- Flush the tube *after* feeding with 5–10 ml of lukewarm tap water.
Maintenance and removal

- Once a day, the neck bandage and sterile dressing should be removed and the stoma cleaned using cotton wool or gauze swabs soaked in 4% chlorhexidine gluconate or 10% povidone–iodine. If oozing of purulent liquid suggests infection, an antibiotic ointment can be applied. A new sterile dressing is applied and the neck bandage re-placed.
- The tube can be removed when it is no longer required; there is no minimum length of time the tube must have been in place.
- To remove the tube, take off the dressing, remove the suture and pull the tube out.
- The stoma site will close rapidly once the tube is removed.

Potential complications

- Infection of the stoma site
- Oesophageal stricture formation (rare)
- Fistula formation (rare)

Further information on assisted feeding can be found in the BSAVA Manual of Canine and Feline Gastroenterology and BSAVA Manual of Canine and Feline Rehabilitation, Supportive and Palliative Care.

Ophthalmic examination

See also Fluorescein test; Schirmer tear test

Indications/Use

- Examination in cases of suspected ocular disease
- Testing vision

Equipment

- Bright focal light source, such as a penlight torch
- Direct ophthalmoscope
- Hand lens (28–30 dioptres (D))
- Topical local anaesthetic
- Mydriatic (e.g. tropicamide)
- Cotton wool balls
- An indirect goniolens
- 0.9% saline
- Tonometer such as a Schiøtz tonometer or TonoPen

Patient preparation and position

- This test is performed in the conscious animal without sedation.
- Position the animal in a standing or sitting position.
Examination with normal illumination and without instruments

1. Begin by observing the patient from a distance.
   i. Assess for evidence of pain.
   ii. Evaluate the blink rate, checking for blepharospasm and photophobia.
   iii. Assess orbital and periocular conformation. Note particularly any asymmetry – by assessing the orbit, the globe size and position – and any strabismus.
   iv. Examine the conformation of the upper and lower eyelids and the position of the nictitating membrane.
   v. Note the presence and nature of any discharge.

2. Now handle the patient.
   i. Examine the lids and conjunctiva.
      a. Retract the upper lid and examine the dorsal bulbar conjunctiva; evert the lid margin and check the upper palpebral conjunctiva and upper lacrimal punctum.
      b. Then apply pressure to the globe through the upper lid and protrude the nictitating membrane, checking its leading edge, its alignment and its outer surface.
      c. Retract the lower lid and observe the lower lacrimal punctum and the conjunctiva as it enters the fornix.
   ii. Check the corneal reflex (the smoothness and sharpness of the reflection of a light on the corneal surface).
   iii. Note the resting pupil size in normal room light.
   iv. Attempt to repel the globes into the orbits by pressing gently through the upper eyelids.

Testing vision

- The menace response is tested by directing a finger towards the eye and observing a blink or retraction of the head. Do not cause a draught of air to contact the cornea as this may give a false positive result.
- Vision may also be tested using cotton wool balls. Hold the ball high and direct the animal’s attention towards it by making a noise. Then release the ball in the animal’s field of view and watch to see whether the animal follows its path.
- Sighted animals, especially young ones, will readily follow the beam from the ophthalmoscope played on the examination table.
- An obstacle course may be improvised from chairs, waste bins, etc. The animal’s performance may be assessed for both eyes or with one eye covered, and in full light or dim light.

Examination in a darkened room, with focal illumination and magnification

1. Examine the adnexa, paying particular attention to the lid margins and conjunctival surfaces.
2. Examine the cornea, looking for lesions such as irregularities, opacities, vascularization and pigmentation, along with their depth and distribution.
3. Continue with the anterior chamber, noting its depth and any abnormal contents. When examining the anterior chamber; it is important to direct the light beam from various angles and also to look at it from different angles.

4. Examine the iris and pupil margin and then the lens.

5. Test the pupillary light reflexes.
   i. Observe the pupil size in normal room light initially.
   ii. With the lights down, direct a bright light axially through the pupil in a distinct on/off fashion, observing the speed and extent of the pupillary constriction.
   iii. In the ‘swinging flashlight test’, the light is directed at each eye alternately, with the observer noting the direct (in the same eye) and consensual (in the other eye) responses.

**Distant direct ophthalmoscopy**

The room should be darkened, but mydriatic drops are not required. The examination is conducted at arm’s length.

1. Set the ophthalmoscope to +1 to +2 D.

2. Find the tapetal reflex (the green or yellow light reflected from the tapetum) by looking into the pupil horizontally or slightly upwards. Any genuine opacity, such as cataracts, corneal and vitreous opacities, in the path of the tapetal reflex will obscure it and appear black.

3. Lesions can be roughly localized in an anteroposterior direction, by taking advantage of the effect of parallax: as the observer moves to one side, the opacity may also appear to move. The direction of this movement can give an indication as to the location of the opacity.
   - Opacities anterior to the plane of the pupil will appear to move in the opposite direction.
   - Opacities in the plane of the pupil (i.e. anterior cataracts), will remain in the same position.
Close direct ophthalmoscopy

The direct ophthalmoscope is used to examine the fundus. The image is upright and highly magnified, but the field of view is very narrow.

1. Hold the ophthalmoscope vertically, with it touching your own orbital margin. Be close to the patient’s eye, so as to maximize the field of view and eliminate distractions such as the iris. The ophthalmoscope should be set between –1 and +1 D for most patients.
2. Find the optic disc initially, as this is more or less at the posterior pole. Evaluate the disc for size, swelling, colour, blood vessels, etc.
3. Make a systematic examination of the fundus. Evaluate the retina for normal variation or changes in colour, reflectivity, pigment, haemorrhage, oedema and detachment.
4. Finally, focus back through to the anterior eye, as required. Unless the presence of vitreal abnormalities is suspected, it is usually simpler to select a setting of around +10 D to bring the lens into focus, the exact setting depending partly on the distance from the eye. This gives a good view of the lens, but with less magnification than for the fundus. Higher positive lenses of +15 to +20 D are required to examine structures anterior to the lens.

Indirect ophthalmoscopy

When compared to direct ophthalmoscopy, indirect methods give an inverted, reversed image which is less highly magnified but covers a much wider angle of view.

1. Dilate the pupils with tropicamide 1% and allow 20 minutes to achieve maximum effect.
2. A simple and effective form of indirect ophthalmoscopy may be carried out using only a hand lens and a focal light source such as a penlight or transilluminator.
3. Hold the lens between your index finger and thumb, and position it in front of the patient’s eye.
4. Hold the light source against your temple or in front of your nose, making sure that your eye, the light source, the lens and the patient’s pupil all lie on the same axis as far as possible.
5. Examine the whole fundus.

**Gonioscopy**

Gonioscopy is the examination of the iridocorneal angle and ciliary cleft.

1. Apply topical local anaesthetic to the cornea and wait approximately 10 minutes for this to take effect.
2. Apply saline to the concave side of the inverted goniolens. Air bubbles should be avoided, because they will distort light and prevent adequate visualization.
3. Place the goniolens on to the cornea in one quick motion so as not to lose the saline.
4. Using a bright light source, such as a penlight torch or direct ophthalmoscope, examine the entire iridocorneal angle and ciliary cleft systematically.

**Tonometry**

Tonometry is the indirect measurement of the intraocular pressure (IOP). Various methods can be used, including indentation and applanation techniques.

**Indentation tonometry using a Schiøtz tonometer**

1. Apply topical local anaesthetic to the cornea and wait approximately 10 minutes for this to take effect.
2. Restrain the patient with the head and eyes directed upwards and the eyelids held open against the orbital rim. Care must be taken not to press against the globe, because this might falsely elevate the IOP.
3. Rest the footplate of the instrument on the central cornea, with the instrument held vertically.
4. Release the plunger and record the IOP reading on the recording scale.
5. Whilst keeping the tonometer in the same place on the central cornea, repeat three times and take an average of the two closest readings.

**Applanation tonometry using a TonoPen**
1. Apply topical local anaesthetic to the cornea and wait approximately 10 minutes for this to take effect.
2. Restrain the patient with the head and eyes directed forwards and the eyelids held open against the orbital rim. Care must be taken not to press against the globe, because this might falsely elevate the IOP.
3. Place a disposable sterile protective sheath over the tip of the TonoPen.
4. Using the probe tip, gently and repeatedly contact the central area of the cornea.
5. Record the IOP measurement from the digital screen. Many machines will also provide an average reading.

More detail on these procedures and interpretation of the results can be found in the *BSAVA Manual of Small Animal Ophthalmology.*

**Orthopaedic examination**

**Indications**
- Lameness

**Observation**
Take note of the following:
- Difficulty and/or reluctance in getting up or walking
- Failure to take full weight on one or more limbs
- Shifting weight from one leg to the next while standing or sitting
- Posture
- Body shape abnormalities:
  - Focal muscle loss
  - Medial/lateral deviation of distal limbs
  - Limb hyperflexion or hyperextension
  - Angular or rotational deformities
- Evidence of old injuries; in particular, check feet, nails, pads and interdigital skin

**Orogastric intubation** see
- Gastric decompression
Gait examination

- The animal should be observed walking and trotting towards and away from the veterinary surgeon on a firm level surface, as well as in front of or around the veterinary surgeon.
- For most dogs this is best accomplished outdoors with the animal on a lead.
- Cats and very small dogs are best observed walking around the consulting room.

Assess for:
- Thoracic limb lameness:
  - A downward nod of the head as the ‘sound’ limb is placed to the ground
  - Lifting of the head as the lame limb strikes the ground
- Pelvic limb lameness:
  - ‘Hiking up’ of the gluteal region on the lame side during weight bearing
  - ‘Bunny hopping’ gait in bilaterally affected animals.
- Swinging leg lameness:
  - Seen while the affected limb is in flight
  - Characteristic pattern of movement, depending upon the injury
  - For example:
    - Pelvic limb: contracture of the gracilis muscle results in outward rotation of the tarsus, with inward rotation of the foot
    - Thoracic limb: contracture of infraspinatus will cause the foot to swing in a lateral arc during protraction.
- A shortened gait or reluctance to move a joint or joints through the full range of motion
- Abnormal flicking forward of the feet during protraction, e.g. due to reluctance to flex the elbows
- Neurological problems leading to paresis (weakness), ataxia (incoordination), spasticity (stiffness) or hypermetria. These should be distinguished from lameness due to orthopaedic disorders by performing a full neurological examination.

Palpation

Palpation of the axial and appendicular skeleton should follow a set sequence:

i. Palpate the spine, starting at the head and finishing at the tail.
ii. Palpate each limb from top to toe, comparing each leg with its opposite.

Take note of the following:
- Changes in muscles: check for asymmetries of shape due to swelling, atrophy, spasm, contracture or weakness
- Anatomical deformities: check the spatial relationship of normal anatomical structures, particularly bony prominences
- Swellings (may involve bones, joints or soft tissues)
• Joint effusions: best appreciated where the joint capsule is least supported by the periarticular structures:
  – Elbow effusions – caudolateral aspect
  – Carpal effusions – dorsal aspect
  – Interphalangeal effusions – dorsal aspect
  – Stifle effusions – either side of the patellar ligament
  – Tarsal effusions – just in front of the calcaneus.
• Joint thickening
• Pain from bone, muscle, tendons, neural tissue or joints.

Manipulation
Manipulation of the axial and appendicular skeleton should follow a routine sequence:
  i. Flex the lower cervical spine and extend it in the axial plane and to the left and right.
  ii. Flex, extend and rotate the lumbar spine.
  iii. Manipulate each limb proximally to distally.

Assess for:
• Anatomical deformity or displacement (e.g. luxation, subluxation)
• Pain
• Range of movement of entire limb and each joint
• Crepitus
• The integrity of the supporting structures of each joint (collateral, dorsal, palmar, plantar ligaments, plus specialized structures such as cruciate ligaments and patella).

Examination of the anaesthetized or sedated animal
Once a seat of lameness has been identified in the conscious animal, additional information may be obtained under sedation or anaesthesia:

• Some manipulations may be uncomfortable whether or not structures are abnormal (e.g. Ortolani test)
• A joint may be too painful to allow full assessment (see Cranial draw test; Tibial compression test)
• Muscle tension in the conscious dog may mask instability
• Instability may be subtle.

Assessment of the nervous system
An orthopaedic examination should routinely include assessment of deep and superficial pain perception, proprioception in all four limbs, and spinal reflexes in the limbs (see Neurological examination).

More detail on these procedures and interpretation of the results can be found in the BSAVA Manual of Canine and Feline Musculoskeletal Disorders.
Ortolani test

Indications/Use

- To detect hip laxity in the young dog (to support a diagnosis of hip dysplasia)

Not all dogs with hip dysplasia show a positive Ortolani sign. For example, dogs with gross subluxation or luxation of the femoral head, and dogs in which capsular fibrosis has stabilized the hip joint will not show the sign.

Patient preparation and positioning

- May be attempted in the conscious animal but is potentially painful and therefore best performed with the dog heavily sedated or under general anaesthesia.
- The animal may be positioned in lateral or dorsal recumbency. The description below applies to lateral recumbency.

Technique

1. Position the stifle in mild flexion and grasp it with one hand, with the other hand placed on the dorsal aspect of the pelvis to stabilize it.

2. Apply firm pressure to the stifle in a dorsal direction in an attempt to subluxate the hip joint.

3. Whilst maintaining dorsal pressure on the stifle, gently abduct the limb until a ‘click’ or ‘clunk’ is detected.
   - If the dorsal acetabular rim is intact, the femoral head falls abruptly into the acetabulum.
   - In dogs with a poor dorsal acetabular rim, the femoral head appears to slide back into the acetabulum.

4. Whilst maintaining dorsal pressure on the stifle, if the limb is now adducted, re-luxation of the hip will occur.

Results

- The ‘click’ or ‘clunk’ (see Step 3) represents the relocation of the femoral head within the acetabulum. This is the positive Ortolani sign, consistent with hip joint laxity.
Otoscopy

Indications/Use
- To obtain specimens for diagnosis of external ear canal disease
- To treat otitis externa. (For sampling and treating otitis media see Myringotomy)

Equipment
- Otoscope and cones of varying lengths and diameters
- OR a video-otoscope
- Cotton wool
- Haemostats for hair removal (if necessary)
- Sterile microbiology swab and transport medium
- Cotton wool buds
- Microscope slides
- 0.9% sterile saline
- 10 ml syringes
- Kidney dish
- Sprula needle or sterile catheters (e.g. 3.5 Fr tomcat catheter, long polypropylene catheter, 3.5 Fr feeding tube) of length sufficient to pass along the full length of the external canal

Patient preparation and positioning
- The animal can be examined standing, sitting or in lateral recumbency.
- Sedation or general anaesthesia may be required.
- Hair is removed, if necessary, by grasping groups of hairs with haemostats and twisting the handle.

Technique
1. Examine the pinnae and entrance to the ear canals with the naked eye.
2. Palpate the ear canal to evaluate pain, thickening and ossification of the ear canal. Note the presence of pus or cerumen. Examine each ear with the otoscope, starting with the normal ear if there is

Oscillometric blood pressure measurement see
- Blood pressure measurement – (b) indirect

More detail on this procedure and interpretation of the results can be found in the BSAVA Manual of Canine and Feline Musculoskeletal Disorders.
one. Apply lateral tension to the ear pinna with fingers to straighten the ear canal as the otoscope is advanced.

3. Visualize the tympanic membrane; in a normal ear this appears as a concave translucent membrane, with a dorsal, white, thick, well vascularized pars flaccida and a ventral semi-transparent pars tensa.

4. If significant pus or cerumen obscures evaluation of the ear canal or tympanic membrane, the ear can be flushed with lukewarm sterile saline. This is drawn into a 5–10 ml syringe attached to the sprula needle or catheter, flushed into the ear canal, and re-aspirated. If possible, this is best performed under otoscopic visualization, with the needle or catheter passed through the otoscope cone.

Sample handling

- Obtain samples for microbiological culture first. These can be collected using a sterile swab inserted through the sterile cone of an otoscope attachment and then placed in transport medium for dispatch to the laboratory.
- To obtain cytology samples, a cotton bud or sterile swab is inserted down to the junction between the vertical and horizontal ear canals. The bud is rotated to collect debris:
  - This may be transferred to a slide with liquid paraffin and a coverslip applied prior to examination with a microscope, using low (4X objective) power
  - A smear can be made by rolling the cotton bud gently on to a microscope slide, then air-dried and stained, as required.

Potential complications

- Rupture of tympanic membrane
- Injury to external ear canal

Further information on video-otoscopy can be found in the *BSAVA Manual of Canine and Feline Endoscopy and Endosurgery.*

Oxygen therapy *see*

- Nasal oxygen administration
Pericardiocentesis

Indications/Use
- Relief of pericardial tamponade
- To obtain a sample of pericardial fluid for diagnostic evaluation

Contraindications
- Suspected coagulopathy
- Active pericardial haemorrhage
- Severe arrhythmia

Equipment
- As required for Aseptic preparation – (a) non-surgical procedures
- Long (5–12.5 cm) wide-bore (14–18 G) over-the-needle catheter
- OR a pericardial drainage catheter set (containing a 9 Fr, 20 cm long catheter with six distal side holes, an 18 G, 7 cm long introduction needle, and a 50 cm guide wire)
- Narrow-gauge cat urinary catheter
- Local anaesthetic
- No. 11 or 15 scalpel
- 3-way tap
- Extension tubing
- 50–60 ml syringe
- Measuring bowl or jug
- EDTA and sterile plain collection tubes
- Microscope slides
- Suture materials or tissue glue
- ECG equipment

Patient preparation and positioning
- Sedation may be required to minimize movement.
- The animal is usually placed in sternal or left lateral recumbency, with pericardiocentesis performed via a right-sided approach.
- The procedure can also be performed using a left-sided approach. Thoracic radiography or ultrasonography may indicate where the heart is most closely associated with the body wall and thus determine the puncture site. Care must be taken, however, to avoid the larger coronary arteries on the left.
- The ECG leads are connected to the patient (see Electrocardiography).
- Aseptic preparation – (a) non-surgical procedures is performed on an area from the 3rd to the 7th rib, in the ventral half of the right chest wall.
Technique

1. The point of needle entry is intercostal space 5 or 6 at the level of the costochondral junction.

2. Infiltrate the skin and subcutaneous tissue with local anaesthetic at this site.

3. Make a small stab incision through the skin with a scalpel blade.

4. If using an over-the-needle-catheter:
   i. Insert the cannula and stylet through the stab incision.
   ii. Slowly advance through the intercostal muscles just cranial to the rib in a slightly cranial dorsal direction until the tip of the needle can be felt scratching against the visceral pericardium.
   iii. A sharp stabbing motion is often required to penetrate the pericardium.
   iv. Pericardial fluid (usually a ‘port wine’-coloured fluid) should appear in the hub.
   v. Withdraw the stylet to leave the cannula in the pericardial sac.

   - The cannula of the over-the-needle-catheter is prone to kinking, usually at the point it passes through the intercostal muscles or as it enters the pericardium.
   - This can be avoided by introducing a narrow-gauge sterile urinary catheter through the cannula into the pericardial sac after withdrawal of the stylet.
   - Fluid can then be drained via this urinary catheter.
If using a pericardial drainage catheter set:
i. Insert the introduction needle through the stab incision and advance the needle as described above.
ii. Pericardial fluid (usually a ‘port wine’-coloured fluid) should appear in the hub.
iii. Insert the guide wire (soft-end) through the needle and into the pericardium.
iv. Remove the needle leaving the guide wire positioned in the pericardium.
v. Thread the catheter over the guide wire, through the skin and intercostal space and into the pericardium.
vi. Remove the guide wire, leaving the catheter in the pericardium.

ECG monitoring throughout the procedure allows identification of arrhythmias caused by the catheter traumatizing the epicardium. If an arrhythmia occurs, the needle should be withdrawn slightly so that it no longer touches the epicardium.

5. Attach the cannula or pericardial catheter to a 3-way tap, extension tubing and 50–60 ml syringe to create a closed drainage system, which can be operated by an assistant.
6. Aspirate the pericardial fluid.
7. If the flow of fluid stops, advance or withdraw the catheter by a few millimetres and re-aspirate, but avoid touching the epicardium.
8. Place 2–3 ml of the pericardial fluid in a plain tube and observe for clot formation, for up to 13 minutes, tilting the tube every 30 seconds. If it clots, the fluid includes fresh blood and the procedure should be stopped. The PCV of the fluid (measured using a microcentrifuge) can also be checked to confirm that it is less than the PCV of peripheral blood.
9. Continue drainage until as much fluid as possible is removed.
Note: it is not necessary to remove all pericardial fluid.
10. After removal of the cannula or pericardial catheter, place a single suture in the skin incision, if required, or close with tissue glue.

Sample handling
- Place the pericardial fluid in an EDTA tube for cytological analysis.
- Also make air-dried smears, especially if the sample is to be posted to an external laboratory.
- If there is a suspicion of infectious pericarditis, submit fluid in a sterile plain tube for bacteriological culture.

Potential complications
- Haemorrhage from the heart is uncommon and, if it occurs, is unlikely to result in catastrophic bleeding. The coronary arteries located on the right epicardium are relatively small, and the right ventricle is under relatively low pressure.
• It is common for arrhythmias to occur as the procedure is performed, usually due to epicardial interference, but they do not usually cause clinically significant consequences.

Management following pericardiocentesis is described in the *BSAVA Manual of Canine and Feline Emergency and Critical Care*. For interpretation of the results of fluid analysis, see the *BSAVA Manual of Canine and Feline Clinical Pathology*.

**Peripheral venous catheters** see Intravenous catheter placement – (a) peripheral veins

**Peritoneal lavage** see Diagnostic peritoneal lavage

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**Platelet count**

**Indications/Use**

• Assessment of platelet numbers

**Equipment**

• Approximately 2 ml of *fresh* venous blood collected into an EDTA collection tube (see Blood sampling – (a) venous)
• Microhaematocrit tube
• Microscope slides
• ‘Spreader’ slide (as for Blood smear preparation)
• Stain (e.g. Diff-Quik)
• Microscope with oil immersion lens
• Immersion oil

**Technique**

1. Check the blood in the EDTA tube for clots. If clots are present, the subsequent count will be inaccurate; take a fresh sample.
2. Make a blood smear and stain with a suitable stain.
3. Examine the feathered edge of the blood smear for platelet clumps. If clumps are present, the subsequent count will be inaccurate; take a fresh sample and repeat 1–3.
4. Count the platelets in 5 high-power fields (1000X, oil-immersion) and calculate the average.
Reference ranges
- Approximately 1 platelet per high-power field is equivalent to 15 x 10⁶ platelets per litre in dogs and cats.
- The normal reference range for platelets in dogs and cats is >200 x 10⁹/l.

Proctoscopy  see
- Endoscopy of the gastrointestinal tract – (b) lower

Prostatic wash

Indications/Use
To obtain a sample for cytology and bacteriological culture from dogs with:
- Prostatomegaly
- Other abnormalities in shape, symmetry or consistency of the prostate
- Prostatic pain
- Haematuria or pyuria

Equipment
- Sterile male dog urinary catheter
- 4% chlorohexidine gluconate
- Bowl to collect urine
- 500 ml bag of sterile 0.9% saline
- 2 and 5 ml syringes
- EDTA and sterile plain collection tubes
- Microscope slides

Patient preparation and positioning
- Can be performed on the conscious dog, although light sedation is generally preferable.
- The patient is placed in lateral recumbency.
- The prepuce must be cleaned of all debris, using dilute chlorhexidine, and rinsed with warm water.

Technique
1. Introduce a urinary catheter and empty the bladder of urine (see Urethral catheterization).
2. The bladder may also be flushed with saline and emptied again at this point to ensure that all urine has been removed. A sample of the saline flush can be collected and handled as below, for comparison with the prostatic wash to help clarify the site of pathology (bladder versus prostate).
3. Partially withdraw the catheter to the level of the prostate, guiding the catheter per rectum.
4. Massage the prostate per rectum or transabdominally for 1 minute and then inject 1–2 ml of sterile saline into the urinary catheter.

5. After a further minute's massage, aspirate material into the catheter using a 5 ml syringe. It is important to ensure that the contents of the catheter as well as those of the syringe are harvested.

6. Remove the catheter.

**Sample handling**

- For turbid samples, prepare a direct smear (as described in *Fine needle aspiration*).
- It is advisable to place some of the wash into an EDTA tube.
- Also, place a sample in a sterile plain tube for culture if infection is suspected.

**Potential complications**

- Rupture of prostatic abscess
- Rectal perforation
- Ascending urinary tract infection

Details of cytology of prostate samples can be found in the *BSAVA Manual of Canine and Feline Clinical Pathology* and the *BSAVA Manual of Canine and Feline Reproduction and Neonatology.*

**Pulse** *see*

- Cardiorespiratory examination

**Punch biopsy** *see*

- Skin biopsy – punch biopsy
Resting energy requirement

The daily energy requirement of an individual reflects the energy required for basic life processes (often referred to as the resting energy requirement, or RER). This is composed of:

- A small amount of energy used for the assimilation of nutrients
- A variable amount of energy expended for body temperature regulation
- Energy expended in physical activity.

The following equation can be used to calculate the RER in kcal/day for cats or dogs:

\[
RER = 70 \times \text{bodyweight (kg)}^{0.75}
\]

Alternatively, for animals over 2 kg the following equation can be used:

\[
RER = 30 \times \text{bodyweight (kg)} + 70
\]

Note: To convert kcal to kilojoules (kJ) multiply by 4.185

In general, for very sick patients that have not been eating well or have been unable to hold food down, the goal of feeding should be to reach the animal’s RER for calories.
- For most cats, the RER is all they need, even when they are under severe metabolic stress.
- Dogs may require additional calories (e.g. 1.25–1.5 x RER) when under severe metabolic stress (e.g. burns), but under most circumstances meeting their RER is sufficient.

Feeding excessive calories to critical patients may cause a number of untoward effects, including gastrointestinal problems, electrolyte disturbances, hyperglycaemia and hepatic dysfunction. The amount fed to a given patient can always be increased if that patient experiences weight loss.

For further information on energy requirements for sick patients, see the BSAVA Manual of Canine and Feline Rehabilitation, Supportive and Palliative Care.
Retrograde urethrography/
vaginourethrography

Indications/Use
- Haematuria
- Dysuria
- Stranguria
- Urinary incontinence
- Urethral discharge
- Urethral trauma

Equipment
- Water-soluble, iodine-containing contrast medium (150–200 mg iodine/ml; more concentrated solutions can be diluted with normal saline). This contrast can be mixed with an equal volume of sterile jelly (e.g. K-Y) to produce a more viscous medium and better urethral distension. This mixture should be made up well in advance, since small air bubbles introduced at mixing can mimic genuine filling defects due to calculi. It should be stored in a dark place as iodinated media degrade in light.
- Local anaesthetic
- Wide-bore male dog urinary catheters of appropriate size for male dogs or tomcat catheters of appropriate size for male cats. Catheters should fit snugly to limit leakage of contrast out of the urethra (see Urethral catheterization)
- Foley urinary catheters of appropriate size for bitches and queens (see Urethral catheterization)

Patient preparation and positioning
- Urethrography is best performed under general anaesthesia, especially in females.
- The patient is positioned for Urethral catheterization.

Technique

Retrograde urethrography (males)
1. The urethral area should be assessed on plain radiographs for evidence of soft tissue swelling or the presence of radiopaque calculi prior to performing contrast studies. A pneumocystogram can be performed, to produce back pressure against which to distend the urethra, but this is not advised in cases of suspected urethral or bladder rupture.
2. Catheterize the urethra with the widest catheter possible, prefilled with saline to avoid producing air bubbles.
3. Position the tip of the catheter distal to the area under investigation.
4. Hold the preputial sheath or penis tightly around the catheter. Local anaesthetic can be instilled into the urethral catheter prior to contrast medium to reduce muscle spasm.
5. Inject 1 ml/kg of the diluted contrast medium as a bolus.
6. Take radiographs immediately after injecting. Lateral views are usually most helpful. Ventrodorsal views can give extra information about the prostatic urethra: slight obliquity from the ventrodorsal position will avoid superimposition of the urethra over itself or over bony structures.

**Retrograde vaginourethrography (females)**

1. The urethral area should be assessed on plain radiographs for evidence of soft tissue swelling or the presence of radiopaque calculi prior to performing contrast studies. A pneumocystogram may be performed first, especially if the bladder is not full.
2. Remove the Foley catheter and cut the tip off beyond the inflatable bulb to prevent it passing too far into the vagina; in cats, a non-cuffed catheter is often used.
3. Prefill the catheter with saline and place the catheter tip just inside the vulval lips, holding the vulva closed around the catheter using, for example, gentle bowel clamps or tongue forceps.
4. Inflate the bulb of the catheter.
5. Inject a little local anaesthetic and then the diluted water-soluble contrast medium. Dose rate is up to 1 ml/kg over 5–10 seconds. Take care to avoid vaginal rupture.
6. Take radiographs immediately after injecting. Lateral views are standard but an oblique ventrodorsal view (to avoid superimposition of the vagina and urethra) may also be helpful.

**Potential complications**

- Urinary tract infection
- Urethral rupture

Further details of these procedures and their interpretation can be found in the *BSAVA Manual of Canine and Feline Abdominal Imaging* and the *BSAVA Manual of Canine and Feline Nephrology and Urology.*

**Rhininoscopy**

**Indications/Use**

- Investigating clinical signs of nasal disease
- Investigating dysphagia, head shaking, pawing at the nose and halitosis in the absence of dental disease
- Foreign body removal
- Collecting samples for histology, cytology and microbiology

**Contraindications**

- Inadequate investigation prior to endoscopy
- Coagulopathy
Equipment

- Caudal rhinoscopy: *flexible* endoscope (diameter 3.5–6 mm)
- Anterior rhinoscopy: *flexible* (as above) or *rigid* endoscope (diameter 2.4–2.7 mm, 0–30 degrees) with sheath or cannula to allow fluid irrigation
- Endoscopic viewing equipment
- 1 litre bags of 0.9% saline (pre-warmed to 37°C) and fluid giving set for irrigation
- Large swabs or bandage roll to pack the caudal pharynx
- Mouth gag
- Water-soluble lubricant (e.g. K-Y jelly)
- 7 Fr 20–40 cm grasping forceps
- Spay hook
- Flexible cup-style biopsy forceps that can pass through the biopsy channel of the rigid endoscope or 3–5 mm rigid biopsy forceps
- 20–60 ml syringes
- Sterile kidney dish
- Container with 10% buffered formalin
- Hypodermic needle: 21 or 23 G
- Tissue cassette with foam insert (‘cell-safe’ frames)

Patient preparation and positioning

- **General anaesthesia is essential.** Always place a *cuffed* endotracheal (ET) tube.
- The patient is placed in sternal recumbency, with the head propped up slightly on towels to elevate the mouth and nasal planum for ease of access. The nose can be angled downwards to direct liquid away from the pharynx.
- A mouth gag is essential when performing *caudal* rhinoscopy to keep the mouth open and prevent the patient biting down on to the endoscope.
- The procedure is best performed on a ‘wet table’, due to the large volume of irrigant used. Towels placed on the floor are useful to catch additional spillage.

Care should be exercised if using cold irrigant solutions, especially in small animals. The high surface area and excellent vascular supply of the nares act like a heat sink and can result in hypothermia.

Technique

- It is good practice to examine the nasopharynx and choanae using caudal (posterior) rhinoscopy *before* performing the rostral (anterior) procedure.

**Caudal (posterior, retropharyngeal) rhinoscopy**

1. Insert the flexible endoscope into the mouth.
2. Advance the endoscope caudally and pass the free edge of the soft palate.
3. Retroflex the endoscope into a ‘J’ position behind the soft palate to view the nasopharynx.
4. Withdraw the endoscope rostrally, with the tip still flexed, to advance the tip toward the choanae.

Anterior (rostral) rhinoscopy
1. Place a swab pack in the caudal pharynx and ensure the cuff of the ET tube is inflated.
2. Begin with the normal or less affected side.
3. Coat the shaft of the endoscope sheath/cannula with water-soluble lubricant, being careful not to get any on the lens of the endoscope.
4. Deflect the nasal planum dorsally and introduce the rigid endoscope with its sheath ventromedially (toward the base of the opposite ear) so that it will pass into the ventral meatus of the nasal cavity.
5. With a bag of saline and giving set connected to one of the stopcocks of the cannula, start the flow of fluid.
6. Whilst pointing the endoscope ventrally and medially, advance the endoscope caudally.
7. Pass the endoscope to the level of the posterior nares and nasopharynx.
8. On entering the nasopharynx, just caudal to the posterior nares, the orifice of the eustachian tube can be seen on the lateral wall.
9. Retract the endoscope rostrally and examine the dorsal meatus and ethmoid turbinates.

Biopsy and sample handling
- Samples should *always* be taken, even if no lesions are apparent. In addition to biopsy samples, brushings and swabs may also be taken for cytology or culture.
- It is important to perform *biopsy at multiple sites*, as there is little correlation between visual appearance and specific disease entities.
The operator can use:
- Flexible cup-style biopsy forceps introduced via the biopsy channel of the endoscope
- Rigid biopsy forceps passed alongside the shaft of the endoscope
- Nasal flush (for cytology).

**Flexible cup-style biopsy forceps**
1. Position the tip of the endoscope approximately 3–5 cm rostral to the lesion to be sampled.
2. Insert the forceps through the biopsy channel of the endoscope, with the cup firmly closed, and advance them towards the site of interest.
3. Once the forceps have passed out of the end of the endoscope, open the cup, advance on to the region to be sampled, and close the cups.
4. Withdraw the forceps from the endoscope and gently remove the sample.

**Rigid biopsy forceps**
1. Position the tip of the endoscope approximately 3–5 cm rostral to the lesion to be sampled.
2. Pass the forceps along the dorsal edge of the rhinoscope, opposite the light guide post, to the site of interest.

![Warning]
The tips of the rigid biopsy forceps must not pass beyond the level of the medial canthus of the eyes. Measure the distance between the tip of the rhinarium and the medial canthus and mark it with a piece of sticky tape on the forceps themselves to prevent inadvertent penetration of the cribriform plate.

In smaller patients, where it is not possible to pass the forceps alongside the endoscope, it is useful to measure the position of the lesion by assessing the direction and depth of the tip of the endoscope, and taking samples 'blind'.
3. Once the tips have passed the end of the endoscope, open the tips, advance on to the region to be sampled, and close the tips.
4. Withdraw the forceps and gently remove the sample.

**Nasal flush**
1. Pack the pharynx with swabs. These should be counted and recorded. *Alternatively*, use a single roll of conforming bandage.
2. Fill 60 ml syringes with saline (20 ml syringes for a cat or a dog <5 kg).
3. Place the nozzle of the syringe up one of the animal's nostrils and squeeze the other nostril shut.
4. Holding an empty sterile kidney dish below the nostrils, empty the syringe with moderate force into one nostril.
5. Repeat multiple times for both nostrils.
6. Ensure that all swabs (or the bandage roll) are removed from the pharynx before the patient is recovered from anaesthesia.
Sample handling

- To remove the samples from the biopsy forceps, immerse in 10% buffered formalin. Then rinse the forceps in saline before reinserting them into the endoscope.
- Alternatively, carefully remove samples from the biopsy forceps with a needle and place directly into formalin or lay on the foam insert of a tissue cassette.
- Place tissue samples for bacterial or fungal culture on a bacterial transport swab or in a sterile container.
- Place nasal flush samples into an EDTA tube for cytology and into a sterile plain tube for bacterial culture, if required.
- To minimize cellular degradation, it is preferable to make direct smears of any mucoid material retrieved from a nasal flush and send these to the laboratory unstained.

Foreign body removal

- Cats: tend to be presented with nasopharyngeal foreign bodies that have been coughed up and lodged over the free edge of the soft palate. These can often be removed without the use of an endoscope, using rigid grasping forceps. A spay hook to retract the soft palate rostrally can also aid removal.
- Dogs: more prone to foreign bodies in the nasal cavities. If small, these can be removed under endoscopic guidance using rigid grasping forceps. Larger foreign bodies will require ‘blind’ removal with rigid grasping forceps.
- If the foreign body breaks up and is impossible to remove completely, the nose should be flushed vigorously with sterile saline. The throat pack should be pushed caudally beyond the tip of the soft palate to allow this fluid to drain over the soft palate and into the mouth freely, and the nose lowered to allow drainage.

Potential complications

- Significant mucosal haemorrhage is rare; if it does occur, pressure can be placed over the rostral nares whilst occluding the caudal pharynx with swabs. Alternatively adrenaline can be sprayed into the nares using a urinary catheter or over-the-needle catheter and syringe.
- Penetration of the cribriform plate is uncommon unless significant disease is present and/or the biopsy forceps are passed ‘blind’ or beyond the level of the medial canthus of the eyes.

Further information on endoscopy of the respiratory tract can be found in the BSAVA Manual of Canine and Feline Endoscopy and Endosurgery.

Robert Jones bandage see
- Soft padded bandage
Schirmer tear test

Indications/Use
To quantify aqueous tear production:
• In suspected deficiency
• In animals receiving lacrimotoxic drugs (e.g. sulphonamides)

Equipment
• Schirmer tear test paper
• Stopwatch
• Topical local anaesthetic
• Cotton wool

Patient preparation and positioning
• This test is performed in the conscious animal without sedation.
• The animal is positioned in a standing or sitting position.

Technique
Schirmer I test
This measures aqueous production in an un-anaesthetized eye, and is therefore an indicator of basal and reflex tear production.

1. Holding the other end, place the short end of the strip in the lateral half of the lower conjunctival sac, so that the notch is at the level of the lid margin and the strip is in contact with the lower lid and the cornea.
2. In most cases it is best to hold the lids closed, retaining the strip securely in position.
3. Remove the strip after 1 minute and record the flow of aqueous tears, either by measuring against the template on the box or from the graduations printed on the strip itself.

Results
• Dogs:
  – Mean reference value = 20 mm/min
  – <10 mm/min raises suspicion of keratoconjunctivitis sicca (KCS)
  – <5 mm/min diagnostic for KCS.
• Cats:
  – Mean reference value = 17 mm/min
  – <5 mm/min raises suspicion of KCS.
Schirmer II test
This measures *basal tear production* only, by eliminating reflex tear production induced by contact with the cornea and conjunctiva. Schirmer II is used in human patients, where high reflex tear production may mask low basal tear production; although this situation probably does arise in animals, the test is not commonly performed.

1. Apply one or two drops of local anaesthetic to the eye, blot the excess away with cotton wool, and allow a few minutes to elapse.
2. Perform the test as above.

**Results**
- Dogs: values will be lower than for the Schirmer I test but typically not lower than 50% of the Schirmer I reading (i.e. approximately 10 mm/min in a normal eye).
- Cats: values will be about 80% of the Schirmer I value (i.e. approximately 14 mm/min in a normal eye).

**Potential complications**
- Corneal irritation
- Corneoconjunctival infection

Further information on this technique and its interpretation is given in the BSAVA Manual of Small Animal Ophthalmology.

Seizures – emergency protocol
- An *integrated approach* to patients in status epilepticus can achieve emergency stabilization, therapeutic intervention and diagnostic investigation simultaneously.
- Although immediate anticonvulsant therapy and systemic stabilization are warranted, concurrent history taking, physical examination and diagnostic tests may be useful.

Systemic stabilization
- **A** Airway: intubate if necessary.
- **B** Breathing: administer 100% oxygen via a non-rebreathing mask.
- **C** Circulation: place a large intravenous catheter; once seizures have stopped, start on isotonic saline (10 ml/kg/h).
- Temperature regulation: treat hyperthermia promptly with slow passive cooling if >40°C; stop at 38.5°C to prevent hypothermia.
Drug therapy

1. Diazepam (0.5–1.0 mg/kg i.v. or per rectum) or midazolam (0.2–0.3 mg/kg i.v. or i.m.).

2. If seizures persist, repeat step 1 up to three times every 10 minutes AND begin loading with phenobarbital (loading dose 12 mg/kg i.v. then, if required, two further doses of 3 mg/kg i.v. at 20-minute intervals to a maximum total dose of 18 mg/kg).

   Note: if the animal has been on maintenance phenobarbital, use 2–4 mg/kg i.v. or i.m. as a single dose.

3. If seizures persist, one or more of the following regimes may be used:
   - Propofol (6 mg/kg i.v., given as 1–2 mg/kg boluses to effect, followed by a continuous rate intravenous infusion of propofol at 0.1–0.2 mg/kg/min)
   - Thiopental (10–20 mg/kg, given as 2–4 mg/kg boluses to effect)
   - Continuous rate intravenous infusion of diazepam (0.5 mg/kg/h i.v. diluted in 5% dextrose or 0.9% saline)
   - Pentobarbital (aliquots of 3 mg/kg i.v. every 90 seconds to a maximum of 6 doses).

Diagnostics

- Arterial blood gas: marked metabolic acidosis is common and will resolve following stabilization; however, respiratory acidosis needs immediate treatment.
- Electrolyte analysis: treat immediately with fluid therapy.
- Glucose analysis: if hypoglycaemic, treat with 50% dextrose diluted to 25% (500 mg/kg i.v.) over 15 minutes or treat with oral glucose syrup; if hyperglycaemic, monitor effects of fluid therapy once the seizures have stopped.
- Haematology/serum chemistry: can be affected by seizure activity so may need to repeat 48 hours after stabilization.
- Urinalysis: rule out myoglobinuria and monitor urine output with indwelling catheter.
- ECG: arrhythmias can occur up to 72 hours after the seizures due to myocardial damage.
- Dynamic bile acids.
- Toxicity screen: immediate results will not be available but take blood to submit for cholinesterase levels after stabilization.

   Cerebrospinal fluid sampling: rule out inflammatory disease.
- MRI/CT scan: rule out structural disease.
- Anti-epileptic drug blood levels.

History

- When did the episode start?
- Is there a pre-existing seizure disorder?
- Is the patient on anticonvulsant therapy? If so, what is the dose; when was the last dose; have serum anticonvulsant levels been measured recently?
- Is the patient on any other medications?
- Are there any systemic health problems?
• Has there been a recent change in personality or behaviour?
• Has there been any recent trauma, travel history or toxin exposure?
• Has the patient eaten a meal within the last few hours?

Physical examination
• Complete physical examination.

For further information on treatment of seizures, see the BSAVA Manual of Canine and Feline Emergency and Critical Care.

Semen collection – (a) dogs

Indications/Use
• Artificial insemination
• Investigation of male infertility

Equipment
• Teaser bitch in oestrus
• Semen-collecting device, such as:
  – A single-use, funnel-shaped plastic cone with a sealed end
  – A latex collection cone attached to a clear collection tube
• Semen extender, storage/transport/cryopreservation facilities if insemination is to be delayed

Patient preparation and positioning
• A quiet room is best, with as few people as possible.
• Access to outside runs is needed if the dog prefers to play with the bitch before mounting.
• The teaser bitch must be held firmly during the collection procedure.

Technique
1. With the bitch held firmly, allow the dog to mount the bitch.
2. As soon as the penis protrudes from the prepuce, deflect it back manually. Hold the bulbus glandis in a firm grip, with the enlarged bulbus in the hand, and your fingers constricting at the point of torsion, caudal to the bulbus.
3. Do not attempt collection until full erection has occurred and thrusting movements have ceased.
4. Collect the semen into the collecting device, by sliding the collection device over the erect penis and holding the mouth of the collection device around the penis proximal to the bulbus glandis.
Canine semen usually has 3 distinct fractions:
• 1st fraction: little sperm, slightly cloudy in appearance
• 2nd fraction: sperm-rich, milky appearance
• 3rd fraction: prostatic fluid, clear and colourless in appearance.

5. If the semen is to be frozen: where the ejaculate is well fractionated, only the sperm-rich (2nd) fraction should be collected. If the semen is to be used immediately for artificial insemination, 1–2 ml of prostatic fluid can be allowed into the funnel, and the semen used directly with no other extender.

Semen handling and preservation
• For immediate artificial insemination, dilution is not necessary.
• If semen is to be transported, with a delay to insemination of 2–3 hours or longer, it should be diluted with a semen extender and transported at 4°C. The semen should be carefully rewarmed to 30–35°C prior to insemination.
• For semen cryopreservation, a variety of freezing regimens, extenders and thawing protocols have been published in the literature.

Potential complications
• Contamination with blood if dog thrusts his penis into the collecting device and ruptures small vessels. Blood makes semen evaluation extremely difficult

Semen collection – (b) cats: artificial vagina

Indications/Use
• Artificial insemination
• Investigation of male infertility (although the relationship between semen quality and fertility has not been established for the domestic cat); males with <40% normal spermatozoa may still have good fertility under natural conditions.
• Note: The need for a pre-trained tom means that this method is not usually practical in a clinical situation

Equipment
• Artificial vagina made from the rubber bulb of a Pasteur pipette and a small test tube
• Queen in heat
• Semen extender, storage/transport/cryopreservation facilities if insemination is to be delayed
Patient preparation and positioning
• To accustom the male to the procedure, a 2–3-week training period is usually necessary. This training is only successful in about two-thirds of toms.

Technique
1. Allow the tomcat to mount a queen in heat.
2. Hold the artificial vagina in a position to facilitate intromission by the tom.

Semen handling and preservation
See Semen collection – (c) cats: electroejaculation.

Semen collection – (c) cats: electroejaculation

Indications/Use
• Artificial insemination
• Investigation of male infertility (although the relationship between semen quality and fertility has not been established for the domestic cat; males with <40% normal spermatozoa may still have good fertility under natural conditions)

Equipment
• Rectal probe (1 x 12 cm) of non-toxic plastic, with three electrodes (1.5 mm x 5 cm) mounted longitudinally; the two outer electrodes are linked together, and the central one is of opposite polarity. The electrodes must be affixed tightly to prevent rectal mucosa becoming trapped
• A stimulator (e.g. a custom-made 50 Hz sine-wave electroejaculator with a transformer, capable of delivering voltages between 0 and 30 V, connected to a 220 V source). Commercial electroejaculators are also available
• Lubricant
• 2 ml collection tube
• Semen extender, storage/transport/cryopreservation facilities if insemination is to be delayed

Patient preparation and positioning
• General anaesthesia is required.
• The cat is positioned in lateral recumbency.

Technique
1. Lubricate the probe and insert it carefully approximately 7–9 cm into the rectum, with the electrodes directed ventrally.
2. Extrude the penis by applying gentle pressure at its base.
3. Place the collection tube over the extruded penis.
4. Administer a series of electrical stimuli for each ejaculate. For example:

- A total of 80 stimuli of 2–5 V:
  - 30 stimuli (10 at 2, 3 and 4 V)
  - Then 30 stimuli (10 at 3, 4 and 5 V)
  - Then 20 stimuli (10 at 4 and 5 V).
- The cat is rested for 2–3 minutes between each series
- Stimuli are administered for approximately 1 second from 0 V to the desired voltage, 2–3 seconds at the desired voltage and an abrupt return to 0 V for 2–3 seconds
- For each stimulus the cat responds with rigid extension of the hindlegs, indicating that the electrical stimulus has been adequate. A lack of extension at 2 V or above usually indicates improper positioning of the electrodes, or interference by faeces.

- If two samples are collected within a short time, the second sample will generally have better motility and a higher proportion of normal spermatozoa. More than one semen sample must therefore always be collected for evaluation of fertility.
- A proportion of the spermatozoa will be lost into the urinary bladder. This retrograde flow is normal during tomcat ejaculation, but is perhaps exaggerated by the use of certain sedatives (xylazine, medetomidine).

Semen handling and preservation

- Semen can be stored for 24–48 hours by extending it with a buffer and chilling. A TesT-buffer with 20% egg yolk and 5% glycerol has been used for storing cat semen at 4–5°C.
- For long-time storage cryopreservation is necessary. Cat semen can be frozen in an egg yolk–lactose extender or in TesT-buffer with 20% egg yolk and 5% glycerol.

For further information on semen collection, evaluation and artificial insemination see the *BSAVA Manual of Canine and Feline Reproduction and Neonatology*.

Skin biopsy – punch biopsy

**Indications/Use**

- Obtaining a full-thickness skin sample, especially in cases of diffuse dermatosis. Less useful for ulcers and nodules, as it is difficult to straddle the margin or encompass the lesion.
Contraindications
• Coagulopathy
• History of poor wound healing

Equipment
• 6–8 mm biopsy punch (a 4 mm punch can be used for restricted sites, such as face and feet)
• Curved scissors
• 70% surgical spirit
• Local anaesthetic
• Fine forceps
• Metzenbaum scissors
• No. 11 scalpel
• Container with 10% buffered formalin
• Pieces of card approximately 10 x 10 mm
• Suture materials

Patient preparation and positioning
• Punch biopsy can usually be performed under light sedation, using local anaesthesia.
• General anaesthesia is required for sensitive sites, such as the face and feet, where local anaesthesia is difficult.
• The patient should be positioned with the area to be sampled uppermost.
• Any hair at the biopsy site should be cut with curved scissors, without disturbing the skin surface.
• Minimal skin preparation is required, though the skin should first be wiped with 70% surgical spirit if the sample is to be sent for culture.

Technique
1. Infiltrate the skin and subcutis with 0.5–1 ml of local anaesthetic.
2. Press the biopsy punch firmly against the skin and rotate in one direction.
3. Using fine forceps, lift the base clear and cut it free, using Metzenbaum scissors or a scalpel blade.

Do not grasp the skin sample itself as this will cause crush artefacts.

4. Close the skin defect with a single suture.

Sample handling
• Place the biopsy specimen, subcutis down, on a piece of card. On the other end of the card, draw a circle with an arrow through it to indicate the direction of hair growth.
• Place the biopsy specimen in a container of 10% formalin.
• Specimens for bacterial or fungal culture should be submitted in a sterile container with 2–3 drops of sterile saline to prevent drying in transit.
• Submit each biopsy specimen in a separate, clearly labelled pot to avoid any confusion.

Potential complications
• Bleeding should cease with pressure; continued haemorrhage may suggest a coagulopathy
• Delayed wound healing

For information on skin biopsy and histopathology see the *BSAVA Manual of Canine and Feline Clinical Pathology* and the *BSAVA Manual of Small Animal Dermatology*.

Skin/hair examination

Whilst a complete history and thorough clinical examination are essential for dermatological cases, further diagnostic investigations are of paramount importance in reaching a definitive diagnosis.

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<td>Tape strip</td>
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The following procedures can be performed in part, as required, or all together as part of a complete dermatological evaluation. (See also Fine needle aspiration and Skin biopsy – punch biopsy.)
Patient preparation and positioning

- The procedures below are usually performed in the conscious animal; light sedation may be required in fractious patients.
- The patient may be positioned standing, sitting or in recumbency, depending on the area to be sampled.

Coat brushing for fleas

Equipment

- Flea comb
- Hand lens
- White paper
- Clear adhesive tape
- Microscope slides and coverslips

Technique

1. Stand the patient on top of a large sheet of white paper.
2. Brush the coat briskly with the fingers or a brush for several minutes.
3. Collect the dislodged material from the paper using clear adhesive tape and stick on to a microscope slide.
4. Examine as for a skin scrape.

Wet paper test

1. Stand the patient on top of a large sheet of white paper.
2. Brush the coat briskly with the fingers or a brush for several minutes.
3. Wet a small wad of cotton wool and squeeze out all the excess moisture.
4. Dab the damp cotton wool over the sample left on the white paper.
5. Flea faeces may be confirmed by blood-tinged marks (orange halo) on the cotton wool or paper.

Hair plucking for dermatophyte culture

Equipment

- Dermatophyte test agar
- Haemostats

1. Pluck hairs using haemostats from the advancing edge of a new lesion, and if possible include any skin scale.
2. Place samples on to dermatophyte test agar.
3. Observe the plate daily for 10 days. Pathogenic fungi cause the medium to change colour to bright red. (Contaminants do not cause a colour change until the cultures become aged.)

Coat brushing for dermatophytes: Mackenzie toothbrush technique

The Mackenzie toothbrush technique can be used for screening animals for dermatophytosis.
Procedures in Small Animal Practice

Equipment
- Sterile toothbrush
- Fungal culture medium such as dermatophyte test medium

1. Brush the whole coat of the animal with a new or sterilized toothbrush for several minutes. It is good to brush against the natural flow of hairs.
2. Touch the toothbrush head on to a culture plate several times.
3. Observe the plate daily for 10 days. Pathogenic fungi cause the medium to change colour to bright red. (Contaminants do not cause a colour change until the cultures become aged.)

Wood’s lamp for dermatophytes
The Wood’s lamp can also be used for screening animals for dermatophytosis.

Equipment
- Wood’s lamp
- Magnifying glass

1. The animal should be placed in a darkened room.
2. Switch on the Wood’s lamp and allow it to warm up for 5–10 minutes.
3. Examine the hair coat with the Wood’s lamp. A magnifying glass may also be used.
4. Observe for bright apple-green fluorescence of hairs; 5 minutes should be spent looking, as the fluorescence is sometimes delayed.

- Unfortunately, not all strains of Microsporum canis fluoresce, the number showing positive fluorescence varying between 30 and 90% in published reports.
- Other less common dermatophytes that may also induce fluorescence include M. distortum, M. audouinii, M. equinum and Trichophyton schoenleinii.
- False-positive fluorescence may be observed due to certain topical medications, dead skin scales and some bacteria.

Hair pluck

Equipment
- Haemostats
- Microscope slides or coverslips
- Clear adhesive tape
- Mineral oil (liquid paraffin)

Technique
1. Pluck several hairs, including the roots, using the haemostats.
2. Place the hairs on a piece of clear adhesive tape and affix this to a microscope slide, or mount under a coverslip in liquid paraffin.
3. Examine under a microscope:
   - Examine the tips of the hairs for evidence of breaking and damage, indicated by abrupt, blunt or frayed ends instead of the tapering tips of normal hairs.
   - Examine the roots to assess whether hairs are in anagen (a pronounced bulb present) or telogen (club root with barbs or frayed appearance) phase.
   - The presence of comedones or follicular plugs can also be detected, surrounding the shafts towards the root end of plucked hairs.
   - Examination of the roots of plucked hairs may reveal the presence of *Demodex* or *Cheyletiella* mites, louse eggs or fungal spores (dermatophytosis).

**Adhesive tape impressions**

**Equipment**
- Clippers
- Microscope slides
- Clear adhesive tape
- Stain such as Diff-Quick®

**Technique**
1. Firmly press a piece of clear adhesive tape against an unhaired (or clipped) region of skin.
2. Attach both ends of the tape to a slide, leaving the middle portion free.
3. Stain the tape strip with Diff-Quick® or equivalent. Gently rinse off excess stain. Detach one end of the tape and stick the whole strip down flat on the slide.
4. Blot off excess liquid and perform cytological examination of the tape with a microscope.
5. Alternatively, the tape strip can be examined unstained for ectoparasites.

**Smears of expressed follicular contents**

**Equipment**
- Microscope slides and coverslips
- Mineral oil (liquid paraffin)
- No. 10 scalpel

**Technique**
1. Squeeze the skin to extrude material from hair follicles.
2. Draw a clean microscope slide across the surface to smear this material on to the slide. Alternatively, material may be collected on a scalpel blade and then transferred to a slide.
3. No staining is necessary, although a little mineral oil (liquid paraffin) may be used for coverslip mounting.
4. Examine the slide with a microscope for ectoparasites, especially *Demodex* mites.
Skin scrape

Equipment
- Clippers
- Saline or mounting medium
- No. 10 scalpel blade (blunted)
- Microscope slides and coverslips
- Liquid paraffin (mineral oil)
- 10% potassium hydroxide

Technique
1. Gently clip hair from the area to be sampled, avoiding areas that have been severely traumatized.
2. Apply a drop of liquid paraffin to the skin surface and the scalpel blade.
3. Squeeze the skin between the fingers.
4. Gently scrape the top layer of the skin surface until capillary oozing of blood occurs, and transfer the sample to the slide.
5. Apply a drop of liquid paraffin (mineral oil) or potassium hydroxide to the sample on the slide.

- Liquid paraffin allows immediate examination of slides and identification of live mites, but has the disadvantage of not clearing debris.
- Good separation of keratinocytes and clearing of material is obtained with 10% potassium hydroxide, but this is caustic to the skin itself and kills mites.

6. Place a coverslip over the sample and mounting medium and examine with a microscope, under low and then high power.

Stained smears from pustules or lesions

Equipment
- Hypodermic needles: 21 G
- Microscope slides and coverslips
- Stain such as Diff-Quick®

Technique
1. Rupture an intact pustule with a sterile hypodermic needle, and smear its contents on to a clean microscope slide.
2. Impression smears can also be made from ulcerated lesions or the cut surface of excised lesions.
3. Air-dry the slide.
4. Stain with Diff-Quick® or equivalent, according to the manufacturer's instructions.
5. Perform cytological evaluation using a microscope. Occasionally ectoparasites such as Demodex mites may also be seen.
Skin samples for bacteriological culture

Equipment
- Clippers
- Hypodermic needles: 21 G
- Sterile swab and bacterial transport medium

Technique
1. Samples for bacterial culture should be taken from new lesions or recently ruptured pustules or vesicles and not from old, crusted or excoriated lesions.
2. Do not use any aseptic skin preparation or surgical spirit before sampling.
3. Rupture an intact pustule with a sterile needle and absorb the contents on to a sterile swab.
4. Place the swab into transport medium before sending samples to the laboratory.

Soft padded bandage

Indications
- To control limb oedema and swelling
- To support the limb following surgery

Equipment
- 2.5 cm wide adhesive tape
- Tongue depressor (optional)
- Cast padding or cotton roll
- Conforming gauze bandage
- Outer protective bandage material (e.g. self-adhesive non-adherent bandage)

Patient preparation and positioning
- Sedation or general anaesthesia may be required where the limb is painful or the animal is non-compliant.
- The animal should be placed in lateral recumbency with the affected limb uppermost.

Further details on skin sampling and its interpretation can be found in the BSAVA Manual of Canine and Feline Clinical Pathology and the BSAVA Manual of Small Animal Dermatology.
The affected limb should be supported in a weight-bearing position, and an appropriate dressing placed directly over any wounds or surgical incisions.

**Technique**

1. Place two strips of adhesive tape (stirrups) on the distal limb on *either* the dorsal and palmar/plantar surfaces *or* the medial and lateral surfaces. These tape strips extend beyond the tip of the toes and are stuck to each other or to a tongue depressor.

2. Beginning distally, apply cast padding material or cotton roll up the limb, in a spiral fashion, overlapping the layers by 50%. The bottom of the bandage should be at the level of the nailbeds of digits III and IV.
3. Place conforming gauze snugly over the cotton layer, again overlapping layers by 50%. The conforming gauze should compress the cotton but not create venous stasis. It should not extend proximally beyond the level of the cotton.

4. Separate the tape stirrups, rotate them through 180 degrees, and apply them proximally to the bandage to limit slippage. The pads and nails of the axial digits should remain exposed.

5. Apply an outer protective bandage material to the bandage. This should not be applied tightly: it should be pulled free from the roll, the tension released from the material, and the material simply placed over the gauze.
Robert Jones technique
- The Robert Jones bandage is a heavily padded modification, which provides support for the first aid management of limb fractures.
- It is only suitable for fractures distal to the elbow in the forelimb and distal to the stifle in the hindlimb.
- For fractures of the humerus or femur the distal edge of the Robert Jones bandage acts as a fulcrum, across which the fracture pivots, with the possibility of exacerbating soft tissue injury and fracture displacement.
- One version of the Robert Jones bandage can be achieved by applying three consecutive layers of cast padding material or cotton roll, each of which is compressed with a layer of conforming gauze. The Robert Jones bandage is protected by a final layer of an outer protective bandage material. Bandage materials are applied to the limb as for the soft padded bandage.

Bandage care and maintenance
- Soft padded bandages should be checked every 4 hours for the first 24 hours and at least twice daily thereafter for complications.
- Written instructions should always be given to clients at discharge; owners must understand their responsibility regarding bandage maintenance.
- Exercise restriction is generally indicated.
- The bandaged limb should be monitored for swollen toes, cold toes, wetness, soiling or slippage, and changed if necessary.
- The bandage must be kept clean and dry. A plastic bag may be placed over the foot while the animal is walking outside and then removed when it is indoors.
- If no open wounds are present, the bandage may be changed every 7–10 days. If open wounds are present, the bandage should be changed as needed, sometimes several times daily.

Potential complications
- Venous stasis
- Limb oedema
- Moist dermatitis
- Maceration of skin underlying a wet bandage
- Contamination of wounds
- Pressure necrosis

Specific gravity (urine) see
- Urinalysis
Spica splint

Indications/Use
- Support of the elbow following:
  - Closed reduction of traumatic elbow luxation associated with moderate instability
  - Closed reduction of elbow luxation combined with open repair of collateral ligament injuries
- Support of the shoulder following closed reduction of traumatic lateral shoulder luxation

Equipment
- 2.5 cm wide adhesive tape
- Cast padding
- Conforming gauze bandage
- Resin-impregnated fibreglass cast materials
- Outer protective bandage material (e.g. self-adhesive non-adherent bandage or adhesive bandage)

Patient preparation and positioning
- The animal should be sedated heavily or under general anaesthesia.
- The animal should be placed in lateral recumbency, with the affected limb uppermost and supported in a weight-bearing position.

Technique
1. Apply a light Robert Jones soft padded bandage, excluding the final outer bandage layer, to the entire forelimb and extend this bandage over the dorsal midline to encompass the thorax. Take care not to apply the bandage too tightly to the thorax, so as not to compromise respiration.
2. Follow the manufacturers’ recommendations regarding wetting and handling of the cast material.
3. Apply strips of cast material over the lateral aspect of the bandage. These strips should extend from the distal tip of the bandage to proximally over the dorsal midline to the contralateral scapula. Cast material should be conformed to the contours of the limb and thorax.
4. Secure the cast material to the underlying Robert Jones bandage with an outer protective bandage material.

Splint care and maintenance
- Written instructions should always be given to clients at discharge; owners must understand their responsibility in splint maintenance.
- Exercise restriction must be enforced.
- Spica splints should be checked every 4 hours for the first 24 hours and then at least twice daily thereafter for complications.
The limb should be monitored for swollen toes, cold toes or skin abrasions. The splint should be monitored for wetness, soiling or slippage, and changed if necessary.
The splint must be kept clean and dry. A plastic bag may be placed over the foot while the animal is walking outside and then removed when it is indoors.

### Potential complications
- Dyspnoea
- Venous stasis
- Limb oedema
- Moist dermatitis
- Maceration of skin underlying a wet bandage
- Contamination of wounds
- Pressure necrosis

### Sterile skin preparation see
- Aseptic preparation

### Surgical preparation see
- Aseptic preparation

### Synovial fluid collection see
- Arthrocentesis
Thoracocentesis – needle

Indications/Use

- Rapid stabilization in immediate respiratory distress due to the accumulation of air or fluid in the pleural space
- To obtain samples of pleural fluid for diagnostic evaluation
- See **Thoracostomy tube placement** for alternative techniques to remove a large volume of pleural fluid or very viscous pleural effusions, or where repeated thoracic drainage is required

Equipment

- As for **Aseptic preparation – (a) non-surgical procedures**
- Butterfly needle, 3-way tap and syringe
  OR an over-the-needle catheter, intravenous fluid administration extension tubing, 3-way tap and syringe
  - Needle/catheter sizes: 18–20 G for medium to large dogs (>10 kg); 20–22 G for cats and small dogs (up to 10 kg)
  - Syringe sizes: 10–50 ml dependent on the expected volume of air or fluid within the pleural space
- Measuring bowl or jug to collect pleural fluid
- Local anaesthetic

Patient preparation and positioning

- Manual restraint in combination with sedation should be used, as necessary, to prevent movement of the patient during the procedure, but care should be taken when administering sedatives to a dyspnoeic patient.
- Sternal recumbency is usually the easiest position for the animal and the most efficient for drainage. Alternatively, a sitting position or lateral recumbency may be used.
- **Aseptic preparation – (a) non-surgical procedures** should be performed on an area of skin on the lateral thorax, to include skin within a 15 cm radius of the proposed site of thoracocentesis.
  A skin drape is usually not required.
- Local anaesthetic may be instilled into the subcutaneous tissues and muscles at the proposed site.

Technique

**Butterfly needle**

1. Attach the butterfly needle to a 3-way tap and syringe, with the 3-way tap turned so that it is ‘off’ to the butterfly needle. Hand the 3-way tap and syringe to an assistant.
2. Thoracocentesis is usually performed at the 7th or 8th intercostal space unless radiography or ultrasonography indicates otherwise. The site of needle insertion is:
   • Half way up the thoracic wall if both pleural air and fluid are present
   • In the ventral third of the thorax if only pleural fluid is present
   • In the dorsal third of the thorax if only pleural air is present.

3. Insert the butterfly needle into the thorax at the desired intercostal space along the cranial border of the rib to avoid the intercostal vessels and nerves. Advance the needle slowly in a slightly ventral direction (approximately 45 degrees to the body wall), with the bevel of the needle facing the lung.

4. Once the butterfly needle is felt to enter the pleura, continue to hold and stabilize it within the thorax. The assistant turns the 3-way tap such that gentle suction can be used to drain the pleural space into the attached syringe. No more than 2 ml negative pressure should be applied to the pleural space. The extension tubing attached to the butterfly needle allows the syringe to move independently from the needle in the patient, reducing the risk of lung laceration and minimizing the risk of dislodging the needle from its desired position.

5. The syringe can be emptied via the 3-way tap into a collecting jug or bowl.

6. Drainage is complete when no further fluid or air can be pulled into the syringe or when the veterinary surgeon can feel the lung rubbing against the tip of the needle.

7. The 3-way tap is turned so that it is ‘off’ to the butterfly needle and the needle is withdrawn from the thorax.

8. Drainage of both sides of the thorax is recommended.

9. Radiography should be performed after thoracocentesis to document change and confirm absence of iatrogenic injury.

**Over-the-needle catheter**

1. A non-sterile assistant attaches the 3-way tap to the syringe, but avoids direct contact with the tips/ports of the syringe and 3-way tap.

2. Insert the over-the-needle catheter into the thorax at the desired intercostal space (see above), along the cranial border of the rib to avoid the intercostal vessels and nerves. Advance the catheter slowly in a slightly ventral direction (approximately 45 degrees to the body wall), with the bevel of the needle facing the lung.
3. Once the catheter is felt to enter the pleura, advance it into the thorax over the needle and withdraw the needle.

4. Attach one end of the extension tubing to the catheter and pass the other end to the non-sterile assistant, who attaches it to the 3-way tap and syringe.

5. The assistant turns the 3-way tap such that gentle suction can be used to drain the pleural space into the attached syringe. No more than 2 ml negative pressure should be applied to the pleural space. The extension tubing allows the catheter in the patient to move independently from the syringe, minimizing the risk of dislodging the catheter from its desired position.

6. The syringe can be emptied via the 3-way tap into a collecting jug or bowl.

7. Drainage is complete when no further fluid or air can be pulled into the syringe or when the veterinary surgeon can feel the lung rubbing against the tip of the needle.

8. The 3-way tap is turned so that it is off to the catheter and the catheter is withdrawn from the thorax.

9. Drainage of both sides of the thorax is recommended.

10. Radiography should be performed after thoracocentesis to document change and confirm absence of iatrogenic injury.

Potential complications
- Lung laceration
- Pneumothorax
- Pyothorax
- Haemorrhage

For interpretation of the results of fluid analysis, see the *BSAVA Manual of Canine and Feline Clinical Pathology*. 
Thoracostomy tube placement – (a) trocar tube

Indications/Use

• Removal of air or fluid from the pleural space when frequent or repeated drainage is expected or required
• Medical management of pyothorax
• Stabilization prior to definitive surgical treatment of pleural space disease
• Removal of air and fluid from the pleural space in the immediate postoperative period following thoracic surgery

Contraindications

• Severe respiratory compromise: stabilization with oxygen therapy or thoracocentesis may be required to improve respiration prior to induction of general anaesthesia

Equipment

• As for Aseptic preparation – (b) surgical procedures
• Trocar thoracostomy tube:
  – Diameter should be approximately the width of the mainstem bronchus as seen on thoracic radiographs (14–16 Fr for cats and tiny dogs; 18–24 Fr for small to medium dogs; 26–36 Fr for large to giant breed dogs)
  – Length should be such that the tip sits at the level of the 2nd rib following placement
  – 3 or 4 fenestrations should be made close to the tip of this tube, as necessary, to facilitate drainage of air or fluid along the full length of the intrathoracic section of the tube
• Thoracostomy tube connector
• 3-way tap and two injection caps/bungs
• Gate clamp
• 20–50 ml syringe
• Scalpel
• Measuring bowl or jug to collect pleural fluid
• Local anaesthetic
• Suture materials
• Sterile dressing
• Elizabethan collar

Patient positioning and preparation

• Aseptic preparation – (b) surgical procedures of the lateral thorax, to include an area craniocaudally from the caudal border of the scapula to just caudal to the last rib, and dorsoventrally from the vertebrae to the sternum is required, but the procedure does not need to take place in a designated surgical theatre. If the patient tolerates pre-oxygenation, aseptic preparation is best performed prior to induction of anaesthesia.
• The animal should be maintained in sternal recumbency during preparation but thoracic drain insertion is most easily performed in lateral recumbency.
• General anaesthesia is recommended.
• Local anaesthetic should be applied as a bleb at the proposed sites of insertion into the skin and chest.

Intercostal nerve block can be carried out at the proposed intercostal space of entry and 2 or 3 intercostal spaces cranial and caudal to this.

- The intercostal nerves run just caudal to the ribs in a dorsoventral direction.
- Local anaesthetic should be applied as far dorsally as possible in the tissues caudal to each rib.
- After needle insertion, draw back on the syringe, to ensure that the needle is not within a blood vessel, prior to anaesthetic injection.

**Technique**

1. Make a small skin incision with a blade at approximately the 10th intercostal space, about two-thirds of the way up the thoracic wall. This dorsal insertion site should facilitate removal of air within the dorsal thorax and fluid within the ventral thorax if the tube is placed correctly to pass from the dorsal insertion site cranially and ventrally within the thorax.

2. Insert the thoracostomy tube with the inner trocar into the skin incision and under the skin. Advance it in a cranial and slightly ventral direction to approximately the 8th intercostal space. This creates a subcutaneous tunnel to prevent air tracking from the environment along the tube and into the thorax.

3. Hold the thoracostomy tube and trocar unit perpendicular to the 8th intercostal space and introduce it into the thorax by means of a short, controlled push with the heel of the hand. **Hold the drain firmly close to the tip with the other hand to prevent excessive and uncontrolled entry into the thorax.**

4. Advance the drain a short distance over the trocar to protect the sharp tip.
5. Advance the drain and trocar together in a cranoventral direction, parallel with the thoracic wall to approximately the level of the 2nd rib.

**Alternative**
Placement of the thoracostomy tube under the latissimus dorsi muscle may create a better seal around the tube:

i. Make the initial skin incision directly over a rib.
ii. Tunnel the trocar tip of the thoracostomy tube on to the underling rib. In so doing the trocar tip will have perforated the latissiumus dorsi muscle.
iii. Direct the drain cranially between the rib and the overlying latissimus dorsi muscle.

6. Remove the trocar whilst holding the thoracostomy tube firmly in position.
7. Occlude the tube temporarily with forceps, whilst pre-placing a gate clamp on the tube and attaching the tube to a connector and 3-way tap.
8. Remove the forceps, enabling drainage of the thorax using a syringe attached to the 3-way tap. **Do not apply more than 2 ml of negative pressure to the pleural space.**
9. Place two injection caps/bungs on the 3-way tap and secure the thoracostomy tube to the thoracic wall by means of a Chinese finger-trap suture.
10. Place a sterile dressing over the site of thoracic drain insertion and hold the thoracic drain against the thoracic wall with a thoracic bandage.
11. Radiography should be performed after thoracostomy tube placement to confirm correct positioning within the thorax and to check for complications.
12. Place an Elizabethan collar on the patient to prevent interference with the tube.

**Tube care and maintenance**

- Animals with thoracostomy tubes in place should have constant, ideally continuous, supervision: to ensure security of the tube connections; to observe for changes in respiratory rate and effort; and to prevent self-interference with the drain.
- The pleural cavity should be checked and drained **every 4 hours** to determine an appropriate time for tube removal.
- The dressing and bandage must be changed and all connections checked at least once a day. Thoracic drains should be handled in an aseptic fashion during bandage changes and for drainage of the pleural cavity.
- A non-functional thoracostomy tube should be removed promptly.
Potential complications

- Malpositioning of the tube
- Lung laceration
- Pyothorax
- Pneumothorax
- Accidental removal of the tube
- Collapse of the tube due to excess suction pressure
- Obstruction of the tube with tenacious fluid or as a result of kinking of the drain
- Haemorrhage

Thoracostomy tube placement – (b) small-bore wire-guided

Indications/Use

- As for Thoracostomy tube placement – (a) trocar tube
- Severe lung laceration may be less likely in inexperienced hands with small-bore wire-guided thoracostomy tubes compared to trocar tubes

Contraindications

- Severe respiratory compromise: stabilization with oxygen therapy or thoracocentesis may be required to improve respiration prior to induction of general anaesthesia

Equipment

- As for Aseptic preparation – (a) non-surgical procedures
- 14 G, 20 cm long radiopaque polyurethane catheter with a multi-fenestrated tip and suture wing to facilitate attachment to skin. Supplied in a sterile pack with short extension tubing with syringe tip, 14 and 18 G catheter-over-needle introducers, 60 cm J-tip guide wire, extra suture wings for the catheter, a closed 1-way valve bung, and tethered cap
- 20–50 ml syringe
- Intravenous fluid administration extension tubing attached to a 3-way tap
- Scalpel
- Measuring bowl or jug to collect pleural fluid
- Local anaesthetic
- Suture materials
- Sterile dressing
- Elizabethan collar

Patient positioning and preparation

- Heavy sedation is usually sufficient.
  - General anaesthesia may be employed if required for additional procedures.
If not anaesthetized, local anaesthetic must be applied as a bleb at the proposed site of drain placement.

- **Aseptic preparation** – *(a) non-surgical procedures* of the lateral thorax to include an area at least 15 cm around the proposed site of catheter insertion. A fenestrated drape should be centred over the proposed site of catheter insertion – towel clips are not recommended in unanaesthetized patients.

- The catheter can be placed with the animal in sternal or lateral recumbency depending on the preference of the veterinary surgeon.

**Technique**

1. Make a small skin incision with a blade at the 8th intercostal space, approximately one-third of the way down from the thoracic vertebrae for pneumothorax and approximately one-third of the way up from the sternum in the case of pleural effusions. Note: a subcutaneous tunnel to prevent iatrogenic pneumothorax is not required for this small catheter.

2. Insert the 18 G catheter introducer along the cranial border of the rib to avoid the intercostal vessels and nerves. Advance the introducer slowly in a slightly ventral direction (approximately 45 degrees to the body wall), with the bevel of the needle facing the lung.

3. Once the introducer is felt to enter the pleura, advance the catheter completely into the thorax over the needle and withdraw the needle.

4. Thread the J-wire through the introducer catheter into the thorax. Advance the wire in a cranioventral direction approximately 12–20 cm or until resistance is encountered. **The guide wire should be held at all times when it is partially within the thorax.**

5. Remove the introducer catheter from the thorax over the wire, leaving the wire in place within the thorax.

6. Advance the catheter into the thoracic cavity over the guide wire to the level of the suture wing.

7. Remove the guide wire and attach a 3-way tap and syringe to the catheter. Aspirate fluid or air (depending on the animal’s condition) from the thoracic cavity to check correct positioning of the catheter within the thorax. Detach the syringe from the catheter and close the catheter with a closed 1-way valve bung.

8. Secure the catheter to the skin, using suture material passed through the holes in the suture wing.

9. Apply a sterile non-woven adhesive dressing over the entrance site of the catheter and secure the attached extension tubing to the thorax with a second similar dressing or a bandage passed around the thorax.

10. Radiography should be performed after thoracostomy tube placement to confirm correct positioning within the thorax and to check for complications.

11. An Elizabethan collar should be placed to prevent interference with the drain.
Tube care and maintenance
• As for Thoracostomy tube placement – (a) trocar.

Potential complications
• As for Thoracostomy tube placement – (a) trocar
• A small volume iatrogenic pneumothorax at the time of catheter placement may occur. However, subsequent iatrogenic pneumothorax is unlikely due to the small size of the catheter and relatively atraumatic catheter placement

Tibial compression test

Indications/Use
• To diagnose partial or complete rupture of the cranial cruciate ligament (CCL)
• Note: not all dogs with CCL disease have femorotibial instability that can be detected by this test
• Often used in association with the cranial draw test

Patient preparation and positioning
• Can be performed in the conscious animal. However, if the patient is tense (due to pain or temperament) or if the CCL is only partially torn, sedation or general anaesthesia is required.
• A conscious patient should be restrained in a standing position on three legs, with the affected limb held off the ground.
• Sedated or anaesthetized patients may be positioned in lateral recumbency, with the affected limb uppermost.

Technique
1. Grasp and maintain the distal femur in a fixed position with one hand, placing the thumb over the lateral fabella and the index finger lightly on the tibial crest.
2. Use the other hand to grasp the metatarsal region.
3. Maintain the stifle joint in slight flexion, while slowly flexing the hock.
Results
• Cranial displacement of the tibial crest relative to the femur is suggestive of CCL injury.

More detail on this procedure and interpretation of the results can be found in the BSAVA Manual of Canine and Feline Musculoskeletal Disorders.

Tissue biopsy – needle core

Indications/Use
To obtain samples from:
• Superficial masses that can be palpated well enough to be stabilized
• Relatively deep sites with minimal surgical intervention (e.g. abdominal organs) usually under ultrasound guidance
• As an alternative to fine needle aspiration

Contraindications
• Coagulopathy

Equipment
• As for Aseptic preparation – (a) non-surgical procedures
• 14–20 G core biopsy needle
• Local anaesthetic
• No. 11 or 15 scalpel
• Container with 10% buffered neutral formalin
• Suture materials or tissue glue

Patient preparation and positioning
• The procedure can usually be performed under sedation.
• If the mass to be sampled is superficial, local anaesthetic is infiltrated into the surrounding area.
• Deeper, ultrasound-guided, procedures will normally require general anaesthesia.
• The patient is positioned with the area to be sampled uppermost.
• Aseptic preparation – (a) non-surgical procedures of an area several centimetres either side of the site to be sampled.
**Technique**

1. Make a small stab incision through the skin, using a scalpel blade.
2. Prior to insertion, prepare the biopsy needle by pulling back on the plunger until a firm click is felt, indicating that the needle spring is locked into a ready position.
3. With the stylet fully retracted, so that the specimen notch is completely covered by the cutting cannula, advance the needle into the tissue to be sampled.

![Technique Image 1](image1.png)

4. Advance the stylet with the thumb to expose the specimen notch within the tissue to be sampled.

![Technique Image 2](image2.png)

5. Rotate to and fro to ensure that tissue fills the biopsy notch.
6. Fire the cutting cannula by fully depressing the plunger.

![Technique Image 3](image3.png)

7. Withdraw the biopsy needle.
8. To remove the tissue specimen, pull back on the plunger until a firm click is felt. Push the stylet forward to expose the tissue specimen within the notch.
9. Gently remove the sample with a fine hypodermic needle, or by irrigation with saline into the fixative.

![Technique Image 4](image4.png)

10. Leave the skin incision to heal, or close with a single suture or tissue adhesive.
Sample handling
- Fix solid tissue samples in 10% buffered neutral formalin solution.
- Impression smears of tissue samples can also be made.

Potential complications
- Haemorrhage. Sampling of vascular organs such as the liver, spleen or kidneys should be preceded by an assessment of coagulation status and monitoring for several hours after the procedure.
- Damage to internal viscera.

Tracheostomy
Indications/Use
- Management of upper airway obstruction that is non-responsive to medical management.
- Maintenance of prolonged mechanical ventilation.
- Anaesthesia for certain surgeries of the upper airway and pharynx, where an endotracheal (ET) tube would limit surgical access.

Equipment
- As for Aseptic preparation – (b) surgical procedures
- Tracheostomy tubes:
  - Routine airway maintenance: non-cuffed tracheostomy tube with an inner cannula; outer diameter no more than 75% of the luminal diameter of the trachea.
  - Maintenance of anaesthesia or prolonged mechanical ventilation: tracheostomy tube with an inner cannula and a high-volume low-pressure cuff.
Suture materials
Nylon tape (usually supplied with the tracheostomy tube)
Range of narrow ET tubes and stylet for endotracheal intubation
Soft tissue surgical instrument set

Patient preparation and positioning
Where possible, tracheostomy tubes should be placed under general anaesthesia in a controlled environment.
This most often means placement of an ET tube prior to tracheostomy tube placement. Failure to achieve endotracheal intubation will necessitate immediate tracheal intubation following anaesthetic induction: be prepared to use a stylet to achieve placement of a standard ET tube or a dog urinary catheter instead of an ET tube in cases of severe upper airway obstruction.
The animal should be placed in dorsal recumbency. The neck should be supported in extension by placement of a sandbag underneath it. The forelegs should be pulled caudally and secured on either side of the thorax.
Aseptic preparation – (b) surgical procedures of the ventral neck.

Technique
1. Palpate the larynx and trachea and make an approximately 7 cm (length depends on the size of the animal) midline skin incision, running caudally from the larynx.
2. Separate the sternohyoideus muscles at the midline and retract them laterally to visualize the trachea. The caudal thyroid vein, with small branches on either side, runs along the midline in the fascia between the sternohydoideus muscles. Try to preserve this vessel to avoid unnecessary haemorrhage.

3. Place stay sutures around the tracheal rings just cranial and caudal to the proposed annular ligament incision. These stay sutures allow for stabilization of the trachea when inserting or changing the tracheostomy tube.
4. Make an incision in one of the annular ligaments between the 3rd and 5th tracheal rings. The incision of the annular ligament should not extend more than 50–60% of the diameter of the trachea. The recurrent laryngeal nerves, which look like fine white threads, run laterally either side of the trachea and should be avoided.

5. Insert the tracheostomy tube into the trachea.

6. Appose the skin cranial and caudal to the tube with simple interrupted sutures, allowing a large enough opening for re-placement of the tracheostomy tube into the trachea if necessary.

7. Secure the tracheostomy tube around the neck with nylon tape.

Note that the stay sutures remain around the tracheal rings for postoperative care.

**Tracheostomy tube care**

- 24-hour-care is essential to prevent potentially fatal occlusion of the tube by exudates and airway mucus and to detect tube dislodgement.
- The inner cannula should be removed for cleaning whenever an increased noise or effort associated with breathing is noticed, or every 2 hours initially. The cannula should be cleaned thoroughly using warm water, dried by evaporation and replaced.
- For tracheostomy tubes without an inner cannula, the entire tube should be removed for cleaning. Ideally, a spare tracheostomy tube should be available for immediate placement into the trachea following removal of the dirty tracheostomy tube. The stay sutures placed around the tracheal rings above and below the tracheostomy site should be used to gently bring the trachea to the level of the skin and to open the trachea.
- Humidification: if the inner cannula is repeatedly full of tenacious mucus and exudate, either nebulized air should be provided for periods for the animal to breathe, or 0.1 ml/kg sterile saline should be instilled into the tube every 2 hours (the latter may induce transient coughing).

- Suction: this is not a benign procedure and should be performed only as required. It is more commonly needed in smaller dogs and cats. The patient should be pre-oxygenated for approximately 10 breaths. A sterile suction catheter should be introduced aseptically into the tracheostomy tube and suction applied for no more than 15 seconds, while gently rotating the suction tube. The suction catheter should remain within the tube during suctioning and only be inserted into the vulnerable trachea if absolutely necessary to clear an obstruction distal to the tracheostomy tube.

- The tracheostomy wound should be inspected daily and cleaned with sterile saline-soaked swabs as necessary.

- If the above measures do not relieve breathing difficulty, the whole tube should be changed. This should not be done in the absence of a veterinary surgeon or facilities for endotracheal intubation and administration of oxygen. The patient is pre-oxygenated and the trachea stabilized using the stay sutures placed around the tracheal rings above and below the tracheostomy site. The old tube is removed and a new one inserted rapidly.

**Potential complications**

- Tracheostomy site dermatitis and infection
- Obstruction of the tracheostomy tube
- Tracheostomy tube dislodgement
- Obstruction of the trachea distal to the tracheostomy tube
- Haemorrhage around the tracheostomy site
- Pressure necrosis of the trachea
- Pneumonia
- Damage to the recurrent laryngeal nerves resulting in unilateral or bilateral laryngeal paralysis
- Fatal airway obstruction

## Transtracheal wash

### Indications/Use

- To obtain a sample for cytology and bacteriology from the airways of medium and large-sized dogs
- Can be used where general anaesthesia is a risk to the patient
- Generally yields samples representative of the trachea and primary or (at best) secondary bronchi, although some material from the lower bronchioles and alveoli may be collected
- The upper airway of dogs and cats may also be sampled by **endotracheal wash**
- The lower airways of dogs and cats may be sampled by **bronchoalveolar lavage**
Contraindications
• Compromised respiratory function

Equipment
• As for Aseptic preparation – (a) non-surgical procedures
• Through-the-needle long jugular catheter (19–22 G, 8 inches long for small (<10 kg) dogs; 19 G, 12 or 24 inches long for larger dogs)
• OR 12–14 G over-the-needle catheter and male dog urinary catheter (3–6 Fr)
• Local anaesthetic
• 2 ml syringe and 21 G hypodermic needle
• No. 11 or 15 scalpel
• 500 ml bag of warm 0.9% sterile saline
• 10 ml and 20 ml syringes
• 3-way tap
• Sterile plain and EDTA collection tubes
• Microscope slides
• Dressing materials

Patient preparation and positioning
• Can usually be performed without sedation unless the animal is fractious. If sedation is used, ideally the animal should be awake enough to cough.
• The patient is positioned in sternal recumbency, with the head elevated.
• Aseptic preparation – (a) non-surgical procedures of the skin of the ventral neck from the larynx to the mid-cervical trachea.

Technique
1. The site of catheter placement is either the cricothyroid ligament or between tracheal rings 2 to 5 distal to the larynx.
2. Infiltrate 0.5–1 ml of local anaesthetic into the skin and subcutis over this area.
3. Stabilize the trachea between the thumb and forefinger, and make a small skin incision.
4. Through-the-needle catheter:
   i. Push the catheter through the cricothyroid ligament or between two tracheal rings, with the bevel of the needle facing down.
   ii. Angle the catheter downwards and feed the catheter’s entire length down the trachea. If the catheter does not feed easily, back the needle out of the trachea a short distance, as the tip of the needle may be pressed up against the wall of the trachea.
   iii. Once the full length of the catheter is in the trachea, withdraw the needle and inject approximately 0.5 ml/kg warm sterile saline into the catheter.
Over-the-needle catheter:

i. Place the catheter in the trachea as described above, and remove the stylet.

ii. Thread the male dog urinary catheter through the catheter to approximately the level of the carina (approximately the 4th intercostal space). The tip of the urinary catheter can be cut off, but care must be taken to ensure that it is not left sharp.

iii. Instil approximately 0.5 ml/kg warm sterile saline through the urinary catheter.

5. Immediately aspirate back the material using a 20 ml syringe attached to a 3-way tap.

6. Repeat the injection of saline and aspiration two to three times if required.

7. Coupage and turning the patient may improve yield.

8. Remove the catheter and apply pressure to the region for 2 minutes before covering in a temporary light dressing.

Sample handling

- Submit an aliquot of the sample in a sterile plain tube for culture.
- Place an aliquot in an EDTA tube for cytology.
- Fresh air-dried smears of any flocculent/mucoid material can also be made and submitted to the laboratory for staining.

Potential complications

- Larynx or airway spasm
- Subcutaneous emphysema
- Pneumomediastinum
- Infection at the needle site
- Catheter breakage and aspiration of the catheter into the airway
- Worsening of respiratory status due to stress of the procedure
Details on cytology of upper respiratory tract samples can be found in the *BSAVA Manual of Canine and Feline Clinical Pathology*.

**Tru-cut biopsy** see
- Tissue biopsy – needle core
Urethral catheterization – (a) male dog

Indications/Use
- To collect urine for urinalysis
- To empty the urinary bladder
- To administer radiographic contrast media into the lower urinary tract (see Retrograde urethrography/vaginourethrography)
- Indwelling:
  - To maintain constant, controlled bladder drainage
  - To maintain a patent urethra
  - To monitor urine output
  - To assist with the nursing care of the recumbent patient and of the patient that is unable to urinate voluntarily

Contraindications
- Pre-existing urethral trauma
- Large space-occupying urethral mass or urethral stricture
- Urine collected by catheterization is not suitable for microbiology, as the sample may be contaminated. Cystocentesis is preferred

Equipment
- Catheters

<table>
<thead>
<tr>
<th>Catheter type</th>
<th>Material</th>
<th>Indwelling?</th>
<th>Sizes (Fr)</th>
<th>Length (cm)</th>
</tr>
</thead>
<tbody>
<tr>
<td>Dog catheter</td>
<td>Flexible nylon (polyamide)</td>
<td>No, but can be modified to be indwelling</td>
<td>6–10</td>
<td>50–60</td>
</tr>
<tr>
<td>Silicone Foley</td>
<td>Flexible medical grade silicone</td>
<td>Yes</td>
<td>5–10</td>
<td>30, 55</td>
</tr>
</tbody>
</table>

- Sterile soft gauze swabs
- 4% chlorhexidine gluconate or 10% povidone–iodine
- Gloves
- Sterile aqueous lubricant, e.g. K-Y jelly
- Plain sample pot
- Kidney dish
- 5–10 ml syringe
If the catheter is to be indwelling:
- For silicone Foley catheters, water sufficient to fill balloon
- For standard flexible nylon (polyamide) catheters: suture materials; zinc oxide tape
- Sterile intravenous fluid administration set and empty fluid bag or commercial closed urine collection system, with appropriate adapters for attachment to selected urinary catheter
- Elizabethan collar

**Patient preparation and positioning**
- Male dogs will generally allow urethral catheterization under gentle physical restraint.
- Sedation may be required for fractious patients.
- General anaesthesia may be required for humane reasons, e.g. patient with fractured pelvis.
- The patient should be restrained in lateral recumbency, with the upper leg held away from the prepuce.
- The prepuce should be cleaned with the antiseptic solution, using swabs or a syringe.
- The area around the prepuce can be clipped, especially in long-haired breeds.

**Technique**
1. Remove the catheter from the outer wrapper and cut a feeding sleeve from the inner sterile packaging, to allow easy feeding of the catheter into the urethra using a ‘no touch’ technique. If using a silicone Foley catheter, feed the guide wire up the centre of the catheter.
2. An assistant should grasp the caudal os penis with one hand and retract the prepuce caudally with the other hand, exposing the glans penis.
3. Lubricate the catheter and insert the tip into the urethra.
4. Advance the catheter into the urethra. Resistance may be met: at the os penis, where there is a slight narrowing of the urethra; at the ischial arch; and at the prostate, if enlarged.
5. Once the catheter is inserted to the level of the caudal os penis, the grip on the penis is relaxed to allow further unobstructed passage of the catheter. Angling the penis caudally may straighten the urethra to ease passage. Steady but gentle pressure should overcome any resistance. If the catheter cannot be passed, re-evaluate the catheter size.
6. When the catheter tip enters the bladder and urine appears in the catheter hub, continue to advance an additional 2 cm to ensure adequate length beyond the trigone.
7. If using a silicone Foley, inflate the balloon once the tip of the catheter is in the bladder.
8. Proceed according to reason for catheterization (e.g. drain bladder, collect urine sample, administer contrast agent).
Sample handling
• For urinalysis:
  – Approximately 5 ml is required
  – Samples should be collected into a plain tube.

Indwelling catheters
Note: a silicone Foley catheter is preferred for indwelling use.

1. If using a flexible nylon (polyamide) catheter, place zinc oxide tape around the catheter near to the prepuce, and place sutures between the tape and the prepuce.
2. Attach a sterile intravenous administration set and empty fluid bag to the urinary catheter and maintain as a closed collection system. Alternatively, attach a commercial sterile closed collection urine bag to the catheter: a catheter adapter may be required.
3. Place an Elizabethan collar.

Potential complications
• Trauma to the urethra or urinary bladder
• Iatrogenic urinary tract infection

Urethral catheterization – (b) bitch

Indications/Use
As for Urethral catheterization – (a) male dog

Contraindications
As for Urethral catheterization – (a) male dog

Equipment
• Catheters

<table>
<thead>
<tr>
<th>Catheter type</th>
<th>Material</th>
<th>Indwelling?</th>
<th>Sizes (Fr)</th>
<th>Length (cm)</th>
</tr>
</thead>
<tbody>
<tr>
<td>Dog catheter</td>
<td>Flexible nylon (polyamide)</td>
<td>No, but can be modified to be indwelling</td>
<td>6–10</td>
<td>50–60</td>
</tr>
<tr>
<td>Foley</td>
<td>Flexible medical-grade silicone</td>
<td>Yes</td>
<td>5–10</td>
<td>30, 55</td>
</tr>
<tr>
<td></td>
<td>Teflon-coated latex</td>
<td>Yes</td>
<td>8–16</td>
<td>30–40</td>
</tr>
</tbody>
</table>

• Sterile vaginal speculum and light source
• Sterile soft gauze swabs
• 4% chlorhexidine gluconate or 10% povidone–iodine
• Sterile gloves
Procedures in Small Animal Practice

• Sterile aqueous lubricant, e.g. K-Y jelly
• Plain sample pot
• Kidney dish
• 5–10 ml syringe

If the catheter is to be made indwelling:
• For Foley catheter: water sufficient to fill balloon; guide wire
• For standard flexible nylon (polyamide) catheter: suture materials; zinc oxide tape
• Sterile intravenous fluid administration set and empty fluid bag or commercial closed urine collection system, with appropriate adapters for attachment to selected urinary catheter
• Elizabethan collar

Patient preparation and positioning
• Bitches may require sedation.
• General anaesthesia may be required for humane reasons, e.g. patient with fractured pelvis.
• The patient may be positioned:
  – In dorsal recumbency with the hindlimbs held cranially (for direct visualization)
  – In lateral recumbency (right lateral recumbency for a right-handed operator; left lateral recumbency for a left-handed operator) (for digital palpation)
  – In sternal recumbency with the hindlimbs over the edge of the table.
• Clean the vulva with the antiseptic to remove any discharge and surface dirt.

Technique

Direct visualization of urethral orifice
1. If using a dog catheter, remove it from its outer wrapping and expose the tip only from the inner sleeve.
2. If using a Foley catheter, remove it completely from its packaging. Handle the catheter with sterile gloves. Insert a guide wire into the catheter tip to stiffen it.
3. Insert a speculum into the vestibule, taking care not to enter the ventrally placed clitoral fossa. The slit of the speculum should be positioned ventrally, allowing the raised external urethral orifice to be identified on the floor of the cranial vestibule. Visualization of the external urethral orifice is often made easier if an assistant pulls on the ventral vulva lips to straighten the vestibule.
4. Lubricate the tip of the catheter and insert it into the urethral orifice under direct visualization.
5. Advance the catheter along the urethra and into the bladder.
6. If using a Foley catheter, it may be advantageous to insert the catheter all the way into the bladder with the guide wire still in place. Remove the guide wire and inflate the balloon once the tip of the catheter is in the bladder.
7. Proceed according to reason for catheterization (e.g. drain bladder, collect urine sample, administer contrast agent).
Digital palpation of urethral orifice
If a vaginal speculum is not available the catheter can be inserted ‘blindly’, using digital palpation of the urethral papilla.

Sterile gloves are recommended to prevent iatrogenic urinary tract infection.

1. If using a dog catheter, remove the catheter from its outer wrapping and the inner package in an aseptic fashion.
2. If using a Foley catheter, a guide wire may be used but is not usually required.
3. Place sterile water-soluble lubricant on the catheter and on your index finger.

4. Place this finger into the vestibule and, while gently applying pressure on its floor, move the finger cranially. The urethral papilla is palpated as a slit on a slight ‘bulge’ of mucosa.

5. While applying gentle pressure over the papilla, feed the catheter under the finger and guide it into the urethra with your other hand. If the orifice is missed, the catheter will run past the fingertip.
6. If using a Foley catheter, inflate the balloon once the tip of the catheter is in the bladder.
7. Proceed according to reason for catheterization (e.g. drain bladder, collect urine sample, administer contrast agent).

Sample handling
- For urinalysis:
  – Approximately 5 ml is required
  – Samples should be collected into a plain tube.

Indwelling catheters
As for Urethral catheterization – (a) male dog.

Potential complications
As for Urethral catheterization – (a) male dog
Urethral catheterization – (c) tomcat

**Indications/Use**

As for Urethral catheterization – (a) male dog

**Contraindications**

As for Urethral catheterization – (a) male dog

**Equipment**

- Catheters

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<th>Indwelling?</th>
<th>Sizes (Fr)</th>
<th>Length (cm)</th>
</tr>
</thead>
<tbody>
<tr>
<td>Jackson cat catheter</td>
<td>Flexible nylon</td>
<td>Yes</td>
<td>3, 4</td>
<td>11</td>
</tr>
<tr>
<td>Silicone cat catheter</td>
<td>Medical grade silicone</td>
<td>Yes</td>
<td>3.5</td>
<td>12</td>
</tr>
<tr>
<td>Slippery Sam</td>
<td>PTFE</td>
<td>Yes</td>
<td>3, 3.5</td>
<td>11, 14</td>
</tr>
</tbody>
</table>

- Sterile soft gauze swabs
- 4% chlorhexidine gluconate or 10% povidone–iodine
- Sterile gloves
- Sterile aqueous lubricant, e.g. K-Y jelly
- Plain sample pot
- Kidney dish
- 5–10 ml syringe

If the catheter is to be made indwelling:
- Suture materials; zinc oxide tape
- Sterile intravenous fluid administration set and empty fluid bag or commercial closed urine collection system, with appropriate adapters for attachment to selected urinary catheter
- Elizabethan collar

**Patient preparation and positioning**

- Cats usually require heavy sedation.
- General anaesthesia may be required for humane reasons (e.g. patient with fractured pelvis) or where catheterization is difficult (e.g. urethral obstruction).
- The patient should be restrained in lateral recumbency, with the hindlimbs pulled slightly cranially and the upper leg held away from the prepuce.
- The area around the prepuce may be clipped, especially in long-haired breeds.
- Clean the prepuce with the antiseptic, using swabs or a syringe.
**Technique**

1. Remove the catheter from the outer wrapper.
2. Lubricate the tip of the catheter.
3. Extrude the penis by applying gentle pressure each side of the prepuce with two fingers.
4. Gently introduce the catheter into the urethra.
5. To allow safe advancement of the catheter, the prepuce and penis should be grasped and pulled in a caudal direction to straighten out the penile and membranous urethra.
6. As soon as the catheter tip enters the bladder and urine appears in the catheter hub, advance the catheter 1 cm further.
7. Proceed according to reason for catheterization (e.g. drain bladder, collect urine sample, administer contrast agent).
8. For indwelling catheters, attach a sterile intravenous administration set and empty fluid bag to the urinary catheter and maintain as a closed collection system. **Alternatively**, attach a commercial sterile closed collection urine bag to the catheter: a catheter adapter may be required.

**Sample handling**

- For urinalysis:
  - Approximately 5 ml is required
  - Samples should be collected into a plain tube.

**Indwelling catheters**

See **Urethral catheterization – (a) male dog**.

- Note that a catheter made of silicone or PTFE is preferred for indwelling use; a Foley catheter is not suitable for tomcats.

**Potential complications**

As for **Urethral catheterization – (a) male dog**

**Urethral catheterization – (d) queen**

**Indications/Use**

As for **Urethral catheterization – (a) male dog**

**Contraindications**

As for **Urethral catheterization – (a) male dog**
### Equipment

- Catheters

<table>
<thead>
<tr>
<th>Catheter type</th>
<th>Material</th>
<th>Indwelling?</th>
<th>Sizes (Fr)</th>
<th>Length (cm)</th>
</tr>
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<td>Flexible nylon</td>
<td>Yes</td>
<td>3, 4</td>
<td>11</td>
</tr>
<tr>
<td>Silicone Foley</td>
<td>Flexible medical-grade silicone</td>
<td>Yes</td>
<td>5</td>
<td>30</td>
</tr>
</tbody>
</table>

- Sterile soft gauze swabs
- 4% chlorhexidine gluconate or 10% povidone–iodine
- Sterile gloves
- Sterile aqueous lubricant, e.g. K-Y jelly
- Otoscope (may be required)
- Plain sample pot
- Kidney dish
- 5–10 ml syringe

If the catheter is to be made indwelling:
- Suture materials; zinc oxide tape
- Sterile intravenous fluid administration set and empty fluid bag or commercial closed urine collection system, with appropriate adapters for attachment to selected urinary catheter
- Elizabethan collar

### Patient preparation and positioning

- Cats usually require heavy sedation.
- General anaesthesia may be required for humane reasons, e.g. patient with fractured pelvis.
- The cat should be positioned in lateral recumbency (right lateral recumbency for a right-handed operator; left lateral recumbency for a left-handed operator).
- Clean the vulva with the antiseptic to remove any discharge and surface dirt.

### Technique

1. Remove the catheter from the outer wrapper and cut a feeding sleeve from the inner sterile packaging if present, to allow easy feeding of the catheter into the urethra using a ‘no touch’ technique. Alternatively, handle the catheter with sterile gloves.
2. Lubricate the tip of the catheter.
3. Grasp the vulval lips with your non-dominant hand, allowing your dominant hand to pass the catheter along the vestibular floor in the midline.
4. Angle the catheter ventrally, placing gentle pressure until it slips into the urethra.

- The anatomy of the queen is such that the external urethral orifice is found as a depression on the vaginal floor. This allows 'blind' urethral catheterization.
- If 'blind' catheterization fails, an otoscope may be used as a vaginoscope to identify the external urethral orifice.

5. As soon as the catheter tip enters the bladder and urine appears in the catheter hub, advance the catheter 1 cm further.

7. If using a silicone Foley, inflate the balloon once the tip of the catheter is in the bladder.

8. Proceed according to reason for catheterization (e.g. drain bladder, collect urine sample, administer contrast agent).

9. For indwelling catheters, attach a sterile intravenous administration set and empty fluid bag to the urinary catheter and maintain as a closed collection system. Alternatively, attach a commercial sterile closed collection urine bag to the catheter: a catheter adapter may be required.

Sample handling
- For urinalysis:
  - Approximately 5 ml is required
  - Samples should be collected into a plain tube.

Indwelling catheters
As for Urethral catheterization – (a) male dog.

Potential complications
As for Urethral catheterization – (a) male dog

Urethral retrograde urohydropulsion – (a) male dog

Indications/Use
- To flush uroliths lodged in the urethra into the bladder, to restore urethral patency. Uroliths flushed into the bladder lumen can then be removed via cystotomy
- Urethral obstruction with uroliths should be confirmed with radiography prior to performing retrograde urohydropulsion (see Retrograde urethrography/vaginourethrography)
Any animal with urethral obstruction should undergo emergency assessment and stabilization, including intravenous catheterization, blood sampling for an emergency minimum database, and fluid therapy for correction of acid–base and electrolyte abnormalities.

Hyperkalaemia associated with bradycardia should be treated aggressively.

**Equipment**

- As required for Urethral catheterization – (a) male dog; flexible nylon urinary catheters are preferred to Foley catheters for urohydropulsion
- Hypodermic needles: 21 G, 1.5 inch
- 3-way taps
- Intravenous extension tubing
- 20–35 ml syringes
- Sterile intravenous fluid administration set and empty fluid bag or commercial closed urine collection system
- Sterile isotonic fluids (e.g. saline, lactated Ringer’s solution)
- Sterile water-soluble lubricant

**Patient preparation and positioning**

- As required for Urethral catheterization – (a) male dog.
- Sedation or general anaesthesia is recommended for the painful or alert patient, but may not be required for the severely depressed.
- Lateral recumbency is recommended.
- The prepuce should be cleaned and flushed with antiseptic (excluding alcohol).

**Technique**

1. Decompress the bladder if overdistended by cystocentesis, using a needle attached to intravenous extension tubing, a 3-way tap and a syringe. This apparatus permits decompression without repeated puncturing of the bladder wall. The needle is held in position within the bladder, while an assistant withdraws urine.
2. Fill one 10 ml (or 12 ml) syringe with 5 ml saline and another with 5 ml of lubricant. Attach the syringes to a 3-way tap to permit mixing.
3. Insert a lubricated large-bore male dog urinary catheter into the urethra (see Urethral catheterization).
4. Instil 3–8 ml of the lubricant mixture around the uroliths. The tip of the catheter should remain distal to the uroliths. Never attempt to force uroliths retrograde with the tip of the catheter.
5. Insert a gloved index finger into the rectum and occlude the urethral lumen by compressing the urethra against the floor of the bony pelvis.
6. With a moistened gauze swab, occlude the distal urethra by compressing the distal tip of the penis around the urinary catheter.

7. Fill a large syringe with sterile isotonic solution. As a guide, the normal bladder will accommodate approximately 7–11 ml/kg bodyweight, but this volume is most often not required.

8. Attach the syringe to the urinary catheter.

9. Push sterile isotonic solution into the urethra, with the goal of dilating the urethral lumen around the uroliths.

10. Once the urethra is dilated, immediately release digital compression of the pelvic urethra.

11. Continue flushing fluid through the urinary catheter and urethral lumen to propel uroliths into the urinary bladder. Repeated occlusion of the pelvic urethra and flushing of the urethra may be required. Use caution not to overdistend the bladder lumen with fluid. Palpate the bladder regularly to check for overdistension and repeat bladder decompression if required.

12. Confirm successful retrograde urohydropulsion of uroliths into the bladder by retrograde urethrography.

13. If the animal is not taken to surgery immediately for removal of uroliths from the bladder, placement of an indwelling urethral catheter is recommended pending surgery, to maintain urine flow.

Potential complications
- Urethral rupture (rare)
- Urinary tract infection

Further information on the management of urolithiasis can be found in the *BSAVA Manual of Canine and Feline Nephrology and Urology*.

**Urethral retrograde urohydropulsion – (b) tomcat**

**Indications**
- Urethral obstruction:
  - Gentle massage of the penis between the thumb and forefinger, with extremely gentle pressure to the bladder may relieve the obstruction; if not relieved immediately, retrograde urohydropulsion should be attempted
  - Radiography may be performed to check for uroliths but, as radiolucent urethral plugs are much more common than uroliths in cats, it is not seen as a prerequisite
  - Urethral plugs or uroliths lodged in the urethra can be flushed into the bladder, to restore urethral patency. Uroliths can then be removed by cystotomy
Any animal with urethral obstruction should undergo emergency assessment and stabilization, including intravenous catheterization, blood sampling for an emergency minimum database, and fluid therapy for correction of acid–base and electrolyte abnormalities.

Hyperkalaemia associated with bradycardia should be treated aggressively.

Equipment

- A selection of urinary catheters of varying sizes (3–5 Fr) including polypropylene or silicone tomcat catheters, ophthalmic lacrimal duct flush cannulas, red rubber urinary catheters, and over-the-needle intravenous catheters (23 G) without their stylet. In large cats, the tomcat catheter may not reach the bladder: a 3.5 Fr rubber urinary catheter or paediatric feeding tube is recommended for longer-term catheterization
- 3-way taps
- Intravenous extension tubing
- 10–20 ml syringes
- Sterile intravenous fluid administration set and empty fluid bag or commercial closed urine collection system, with appropriate adapters for attachment to selected urinary catheter
- Sterile isotonic fluids (e.g. saline, lactated Ringer's solution)
- Sterile water-soluble lubricant
- Elizabethan collar

Patient preparation and positioning

- General anaesthesia is most often recommended.
- Sedation should not be prolonged because of fluid, electrolyte and acid–base abnormalities.
- Positioning is as for Urethral catheterization – (c) tomcat.
- The prepuce is cleaned with antiseptic (excluding alcohol) and rinsed with warm water.

Technique

1. Decompress the bladder by cystocentesis if overdistended, using a needle attached to intravenous extension tubing, a 3-way tap and a syringe. This apparatus permits decompression without repeated puncturing of the bladder wall. The needle is held in position within the bladder, while an assistant withdraws urine. Remove as much urine as possible to avoid leakage.
2. Perform urethral catheterization – (c) tomcat steps 1 to 5. Flush the urethra while advancing the catheter. Gently twisting the catheter can also sometimes aid its passage. If catheterization is unsuccessful, try different/smaller types of catheter.
3. Once the catheter has been passed into the bladder, empty the bladder by attaching a syringe to the catheter.
4. Collect urine samples for **urinalysis** and culture as required. A **cystocentesis** sample is preferred for culture.

5. Flush the bladder multiple times slowly with warm sterile saline until clear, emptying the bladder each time.

**Indwelling catheters**

The bladder should remain catheterized if:

- Relief of the obstruction was difficult
- The urine stream is small
- The bladder was overly distended and detrusor function may be questionable
- The patient is uraemic or markedly azotaemic, and diuresis is necessary
- Post-obstructive diuresis is likely and measurement of urine output is necessary in the immediate post-obstructive phase.

1. Advance the catheter into the bladder until urine appears.
2. Then advance the catheter at least 1 cm further.
3. Secure the catheter to the patient by placing an adhesive tape butterfly around the catheter at the level of the prepuce and suturing it to the prepuce. **Alternatively**, for cat catheters with suture collars, suture the prepuce to the suture collar using the holes provided.
4. Attach a sterile intravenous administration set and empty fluid bag to the urinary catheter and maintain as a closed collection system. **Alternatively**, attach a commercial sterile closed collection urine bag to the catheter: a catheter adapter may be required. Tape the tubing of the closed collection system to the tail for additional security, to prevent accidental catheter removal.
5. For cats with feline lower urinary tract disease the urinary catheter must usually remain in place for at least 2 days until medical management has taken effect.
6. Place an Elizabethan collar.

**Potential complications**

- Urethral rupture; this should be minimized by gentle technique with adequate lubrication and patience
- Inability to pass a urethral catheter into the bladder. In this situation placement of a cystostomy tube and medical management to decrease urethral spasm and relax the urethra should be considered. Perineal urethrostomy may be required as a salvage procedure
- Urinary tract infection
- Accidental removal of an indwelling urinary catheter
- Accidental disruption of an indwelling urinary catheter, leaving the catheter tip within the bladder

Further information on urolithiasis and its treatment can be found in the *BSAVA Manual of Canine and Feline Nephrology and Urology.*
Urinalysis

Indications/Use
- To obtain information from urine samples

Equipment
- Urine sample in a plain container, obtained by cystocentesis or urethral catheterization or by free catch
- Refractometer
- Dipsticks and results chart
- Distilled water
- Syringe
- Centrifuge
- Pipette, e.g. plastic disposable
- Microscope slides and coverslips
- Gloves
- Microscope

Specific gravity
Urine specific gravity (SG) should be measured using a refractometer. Urine SG can be determined by some dipsticks but this is very unreliable and not recommended.

Technique
1. Open the prism cover.
2. Check calibration of the refractometer using distilled water. This should read 1.000; if it does not, recalibrate as follows:
   i. Unscrew the fixation nut of the calibration screw.
   ii. Turn the calibration screw downwards to bring the scale up or unscrew to bring the scale down until it reads 1.000.
   iii. Reaffix the calibration nut.
3. To view, hold the refractometer horizontally in the direction of a good light source, preferably a natural light source.

4. Wearing gloves, place 1–2 drops of urine on the face of the lower prism and view through the eyepiece. Note where the colour boundary line is and read against the specific gravity scale.
5. After use, clean the prism and cover carefully with a soft wet cloth or damp lens wipe.

### Dipstick tests

These constitute a qualitative to semiquantitative method of monitoring major chemicals of interest in the urine. The dipsticks are designed for monitoring constituents in human urine; therefore, some of the tests are not suitable for use with animal urine, i.e. SG, nitrite, leucocyte esterase activity (WBC) and urobilinogen.

#### Technique

- Only fresh, in-date sticks should be used. Dipsticks should be stored in the original tightly capped container (the lid contains dessicant).
- Fresh urine should be used.

1. Do not touch the test pads with fingers.
2. Dip the urine test strip in fresh urine and tap off any excess.
3. Alternatively, use a syringe to place drops of urine on each test pad.
4. Check the colour changes at the indicated times.
5. Compare the strip with the test chart provided.
Urine sediment analysis

This is perhaps the most important component of a complete routine urinalysis. A suggested method for preparation and analysis is as follows:

1. Use fresh urine or refrigerated urine that has been warmed to room temperature.
2. Mix the sample well but gently, to prevent destruction of any casts.
3. Place a constant volume (10, 5 or 3 ml; often 3 ml is used for cat urine), in a conical centrifuge tube.
4. Gently centrifuge the urine (approximately 1000 rpm for 5 minutes, though will vary with the type of centrifuge used). If a smaller volume of urine (i.e. <3 ml) is used with high speed centrifugation, the amount of sediment obtained may be too small, especially if the urine is very dilute. The high speed may also damage cells and destroy casts.
5. Decant the supernatant (this can be used in the refractometer for SG analysis – see above).
6. A few drops of liquid will remain in the tube. Sediment is mixed in this to resuspend it, either by gently tapping the tube or pipetting up and down.
7. If a specialized urine stain, such as Sedistain, is being used, add 1 drop to the sediment and mix.
8. Place a drop of sediment on a clean microscope slide and place a coverslip over it, avoiding air bubbles.
9. Place the slide on the microscope stage and scan the whole area on low power (10X objective). If unstained sediment is being used, lower the condenser until there is good resolution.
10. Report the presence of crystals at this magnification.
11. Change the objective to high power (40X) and rescan the area. Count and record the numbers of erythrocytes, leucocytes and epithelial cells, casts, crystals and bacteria seen per high power field. It is advisable to count in 5–10 different fields and then calculate an average unless there are very large numbers seen.
12. Report other constituents, e.g. spermatozoa, mucus strands, yeast, fungi, nematode eggs, fat droplets.
13. For further identification of cell types or to differentiate bacteria from particles, use the 100X oil immersion objective. Distinguish fat and air bubbles by fine focusing and moving the condenser. Distinguish Brownian motion of particles from motile bacteria at 100X power.

Information on interpretation of urinalysis, and illustrations of sediment inclusions, can be found in the BSAVA Manual of Canine and Feline Clinical Pathology.
Urine dipstick tests see

• Urinalysis

Urine sampling see

• Cystocentesis
• Urethral catheterization

Urine sediment see

• Urinalysis

Urine specific gravity see

• Urinalysis
**Velpeau sling**

**Indications/Use**
- To hold the shoulder, elbow and carpus in flexion, supporting the forelimb in a non-weight-bearing position
- Immobilizes the shoulder to promote healing of shoulder and scapula injuries, including traumatic medial shoulder luxation and minimally displaced scapular fractures

**Equipment**
- Padded bandage material or cast padding
- Conforming gauze bandage
- Outer protective bandaging material, e.g. self-adhesive non-adherent bandage

**Patient preparation and positioning**
- Manual support, with the animal in a standing position on three legs, is often all that is required.
- Sedation or general anaesthesia may be required for a non-compliant animal.

**Technique**

1. Lightly pad the antebrachium with a padded bandage material.

2. Secure the padded bandage material to the antebrachium with conforming gauze bandage, working in a medial to dorsal to lateral direction.
3. Gently flex the forelimb. Pass the conforming gauze bandage from the medial aspect of the antebrachium, across the lateral aspects of the humerus and scapula, to the dorsal aspect of the thorax.

4. Then pass the conforming gauze bandage around the thorax, just caudal to the contralateral elbow, and back around the flexed forelimb and thorax. This secures the carpus, elbow and shoulder in flexed positions.

5. Apply several more layers of conforming bandage to incorporate the flexed limb and thorax. Take care to pass the bandage around the cranial aspect of the carpus to prevent the dog from stepping out of the sling.

6. Apply an outer protective bandage to the sling.

- The technique above has been described as a ‘non-weight-bearing’ sling and may be appropriate for traumatic lateral shoulder luxations. However, velpeau slings may promote reluxation of unstable traumatic lateral shoulder luxations.
- In the case of medial shoulder luxations, adduction of the humerus against the thoracic wall is most important to give external rotation of the shoulder. Therefore, it may be preferable to bandage the humerus against the thoracic wall prior to incorporating the rest of the flexed limb in the bandage.
Sling maintenance

- The sling is maintained for 2–6 weeks for traumatic shoulder luxations managed conservatively.
- Velpeau slings should be checked every 4 hours for the first 24 hours and at least twice daily thereafter for complications.

Potential complications

- If applied too tightly potential complications include:
  - Limb swelling due to venous stasis
  - Irritation of the skin, especially of the contralateral axillary region
  - Pressure necrosis of the soft tissues
  - Excessive flexion of the carpus, resulting in discomfort and/or trauma to the antebrachium from the claws
- If the sling becomes wet, moist dermatitis and soft tissue maceration may occur
- Velpeau sling loosening results in the animal stepping out of the sling
Water deprivation test

Indications/Use
Diagnostic aid in cases of:
- Central diabetes insipidus
- Primary nephrogenic diabetes insipidus
- Primary (psychogenic) polydipsia

Contraindications
- Dehydration
- Azotaemia
- Pyometra
- Pyelonephritis
- Hypercalcaemia
- Hyperadrenocorticism

- The water deprivation test should be performed only after all other causes of polyuria and polydipsia have been ruled out, limiting the differential diagnoses to central diabetes insipidus, primary nephrogenic diabetes insipidus and psychogenic polydipsia.
- Failure to recognize the more common polyuric syndromes such as pyometra, pyelonephritis, early renal insufficiency, hypercalcaemia or hyperadrenocorticism may lead to an incorrect or inconclusive diagnosis and use of the water deprivation test in these patients may be dangerous.

Equipment
- Urinary catheter
- Electronic scales
- Measuring vessel for water
- Refractometer
- Intravenous synthetic desmopressin (DDAVP)
- 2 ml syringes
- Hypodermic needles: 21 G, ¾ to 1 inch

Technique
It is recommended that the water deprivation test be performed in three consecutive stages:

1. Gradual water restriction.
2. Followed immediately by abrupt water deprivation.
3. Followed immediately by an antidiuretic hormone (ADH) response test – if necessary.
Stage 1 – Gradual water restriction
To minimize the effects of renal medullary washout on test results, progressive water restriction is recommended before abrupt water deprivation. This is usually carried out by the owner in the home environment.

The owner should:
1. Begin reducing the amount of water provided to the animal 3 days before the abrupt water deprivation test is to be performed in the clinic.
2. During the initial 24 hours, allow the dog or cat twice its normal daily water requirement (120–150 ml/kg) divided into 6–8 small portions.
3. During the next 24 hours, give 80–100 ml/kg.
4. Over the last 24 hours, provide normal maintenance requirements (60–80 ml/kg).
5. During the 3-day period of gradual water restriction, owners should feed dry food and monitor the animal's bodyweight on a daily basis.
6. Owners should also be instructed to observe for any significant decrease in the animal's mentation when performing gradual water deprivation. Should this occur the test should be stopped and veterinary attention sought immediately.

Stage 2 – Abrupt water deprivation
The goal of Stage 2 is to achieve maximal ADH secretion and concentration of urine. This would be expected to occur after a 3–5% loss of bodyweight. This procedure must be carried out in the veterinary clinic and is best started early in the day.

1. Completely empty the animal's bladder (see Urethral catheterization; consider an indwelling urinary catheter, especially in females) and collect the urine.
2. Record the urine specific gravity (SG; see Urinalysis), obtain an exact bodyweight and remove all food and water.
3. At 1–2 hourly intervals, again completely empty the urinary bladder, measure the urine SG and reweigh the animal (to monitor for dehydration).
4. Continue until there is either a 5% loss in bodyweight or the urine SG is >1.030 in dogs or >1.035 in cats. The major difficulty with the water deprivation test is that its duration can never be predicted accurately.

The animal must be monitored for signs of CNS depression. Stop the test immediately if these are seen.

5. Some animals will fail to reach the 5% dehydration endpoint by the end of the working day. In that situation, the patient can be transferred to a facility with overnight care so that monitoring of urine SG and bodyweight can continue overnight. Alternatively,
overnight access to water in maintenance amounts (2.5–3.0 ml/kg/h) can be provided; the following morning, water is once again withdrawn and monitoring continued until a 5% loss of bodyweight or concentrated urine SG is reached.

**Stage 3 – Response to intravenous desmopressin**

If the dog or cat has lost 5% or more of its original bodyweight after water deprivation, but the urine SG remains <1.015, an ADH (desmopressin) response test is performed in the clinic.

1. Provide water in maintenance amounts (2.5–3.0 ml/kg/h) for the duration of this stage.
2. Inject synthetic desmopressin, intravenously:
   - 2.0 µg (micrograms) for dogs <15 kg and cats
   - 4.0 µg (micrograms) for dogs >15 kg.
3. Completely empty the animal’s bladder (see Urethral catheterization; consider an indwelling urinary catheter, especially in females) and collect the urine.
4. Record the urine SG (see Urinalysis) every 30 minutes to 1 hour.
5. Stop the test when the urine SG has risen above 1.015 or the animal shows any signs of CNS depression.
6. Continue this stage for a maximum of 8 hours.
7. Upon completion of the test, water should be offered in maintenance amounts (2.5–3.0 ml/kg/h) for 2–3 hours then provided *ad libitum*.

### Results

<table>
<thead>
<tr>
<th>Disorder</th>
<th>Urine SG prior to test</th>
<th>Urine SG after stage 2</th>
<th>Urine SG after stage 3</th>
</tr>
</thead>
<tbody>
<tr>
<td>Central diabetes insipidus</td>
<td>1.001–1.007</td>
<td>&lt;1.008</td>
<td>Increase to &gt;1.015</td>
</tr>
<tr>
<td>Primary nephrogenic diabetes insipidus</td>
<td>1.001–1.007</td>
<td>&lt;1.008</td>
<td>No change (remains &lt;1.008)</td>
</tr>
<tr>
<td>Primary polydipsia</td>
<td>1.001–1.020</td>
<td>&gt;1.030</td>
<td>No additional increase</td>
</tr>
</tbody>
</table>

### Potential complications

- Severe dehydration has the potential to lead to renal failure
- Rapid rehydration following the end of the test has the potential to result in cerebral oedema and neurological signs

Further information on the investigation of PU/PD is provided in the *BSAVA Manual of Canine and Feline Endocrinology*. 
Whole blood clotting time

**Indications**
- Assessment of secondary haemostasis (intrinsic and common pathways). Note that the WBCT will also be prolonged in severe thrombocytopenia and hypofibrinogenaemia.

**Equipment**
- 5–10 ml glass or 5 ml plastic collecting tube (no anticoagulant)
- Stopwatch or timer

**Technique**
1. Blood is collected from the jugular vein (see Blood sampling – (a) venous).
   - To minimize contamination with tissue factor, collect the first 0.5 ml of blood into a syringe and discard this, but keep the needle in the vein.
   - Draw approximately another 4.5 ml of blood into the same syringe.
2. Place the blood in a glass or plastic tube and start a stopwatch or timer.
3. Keep the tube warm by holding it in the palm of the hand.
4. Tip gently to 90 degrees every 30 seconds until the blood has coagulated.
5. Stop the stopwatch/timer and note the time taken for the blood to clot.

**Reference ranges**
Reported reference intervals are:
- Dogs: 3–13 minutes
- Cats: 8 minutes.

Interpretation of clotting test results is covered in the *BSAVA Manual of Canine and Feline Haematology and Transfusion Medicine.*
Notes
BSAVA Members have access to the electronic edition of the *BSAVA Guide to Procedures in Small Animal Practice* as part of their exclusive package of online resources. Members should visit the website regularly as we will continue to add great new tools in 2010, including video demonstrations of some of the procedures we see in practice.

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BSAVA Guide to Procedures in Small Animal Practice

Edited by Nick Bexfield and Karla Lee

The BSAVA Guide to Procedures in Small Animal Practice provides practical, step-by-step guidance on how to perform the diagnostic and therapeutic procedures commonly performed in small animal veterinary practice. In addition, routine clinical examination of the major body systems, and protocols for the management of selected emergencies, are described.

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Contents
Abdominocentesis; ACTH response test; Anaphylaxis – emergency treatment; Arthrocentesis; Aseptic preparation; Barium contrast media; Barium studies of the gastrointestinal tract; Blood pressure measurement; Blood sampling; Blood smear preparation; Blood transfusion; Bone biopsy – needle; Bone marrow aspiration; Bronchoalveolar lavage; Bronchoscopy; Buccal mucosal bleeding time; Cardiopulmonary-cerebral resuscitation; Cardiorespiratory examination; Cast application; Cerebrospinal fluid sampling; Cranial draw test; Cystocentesis; Dexamethasone suppression tests; Diagnostic peritoneal lavage; Ehmer sling; Elbow luxation – closed reduction; Electrocardiography; Endoscopy of the gastrointestinal tract; Endotracheal wash; Fine needle aspiration; Fluorescein test; Gastric decompression; Gastrostomy tube placement; Haemagglutination test; Hip luxation – closed reduction; Intravenous cannula placement; Intravenous catheter placement; Intraosseous urography; Iodinated contrast media; Myringotomy; Nasal oxygen administration; Naso-oesophageal tube placement; Neurological examination; Oesophagostomy tube placement; Ophthalmic examination; Orthopaedic examination; Ortolani test; Otoscopy; Pericardiocentesis; Platelet count; Prostatic wash; Resting energy requirement; Retrograde urethrogram/vaginourethrography; Rhinoscopy; Schirmer tear test; Seizures – emergency protocol; Semen collection; Skin biopsy – punch biopsy; Skin/hair examination; Soft padded bandage; Spica splint; Thoracentesis – needle; Thoracostomy tube placement; Tibial compression test; Tissue biopsy – needle core; Tracheostomy; Transtracheal wash; Urethral catheterization; Urethral retrograde urohydropulsion; Urinalysis; Velpeau sling; Water deprivation test; Whole blood clotting time

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