THORACIC SKILLS

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Thoracocentesis

Thoracocentesis is both diagnostic and therapeutic. The lateral thorax is clipped and aseptically prepared on one or both sides. If an ultrasound is available, a FAST scan is performed to identify fluid or air location. Supplies are gathered: a needle, butterfly, or IV catheter, collection tubing, a three-way stopcock, and a syringe of appropriate size. Using sterile technique, the needle or catheter is blindly inserted at the 7th to 9th intercostal spaces, or at the space indicated by the ultrasound, with guidance if possible. If pneumothorax is suspected, the needle or catheter should be inserted in the dorsal third of the thorax. For pleural effusion, the needle or catheter should be in the ventral third of the thorax. The needle or catheter should always be advanced off the cranial aspect of the rib and into the pleural space, and needles or butterfly catheters should be directed, bevel side facing the lungs and sharp tip parallel to the chest wall. The collection tubing and three-way stopcock are quickly attached to the needle or catheter, and suction gently applied with the syringe.

Thoracic Drain Placement (Seldinger)

Placing a tube in the thoracic cavity is necessary if large amounts of air or fluid are present. A chest tube should be placed if more than two thoracocenteses are needed in the first few hours of presentation. Chest tubes are commercially available both in large-bore rigid tubes and as Seldinger-placed, flexible, fenestrated catheters. Red rubber feeding tubes can be used as an alternative. Three or four holes must be made large enough for fluid to flow through easily.

To prepare for thoracic drain placement, the skin is clipped and aseptically prepared on the affected side(s). The 7th, 8th, or 9th intercostal space is identified at the junction of the upper one-third and the lower two-thirds of the thorax. This will be the entry point for the thoracic drain. While tunneling is usually preferred to reduce the risk of a pneumothorax around the insertion site, these types of catheters have a tendency to kink if tunneled too much and is generally not recommended.

A lidocaine bleb is injected at the insertion site, and allowed to sit for approximately 5 minutes. A permanent marker can be used to identify the rib space above the point of entry so as not to contaminate the prepared skin. Sterile gloves are donned for this procedure and a small procedure drape such as an eye drape is used to cover the procedure site.

The introducer catheter (an over-the-needle catheter included in the kit) is introduced perpendicular to the skin, between the two ribs and at the site of the lidocaine. Once in the pleural space (often a distinct "pop" is felt), the catheter is advanced off the stylet while being directed cranially. The stylet is removed, and a gloved finger is used to cover the catheter hub while the coiled guidewire is removed from the kit. The guidewire has a soft J-bend in the end, which will prevent damage to lung tissue. Pull the "J" back into the introducer coil, to staighten it, and place the tip into your introducer catheter. Begin to feed the guidewire into the chest, taking note of the marks on the guidewire which are separated at 10cm intervals. Advance as far as desired for the patient size. Adapter wings are included in the kit for use when less than 20cm will be placed in the chest. This type of drain MUST be inserted a minimum of 10cm as there are multiple fenestrations on the tube to the 10cm mark. Once at the desired depth, leave the guidewire in place and keep it stable so it does not push in, or pull out, and remove the coiled wire cover off of the wire, then remove the introducer catheter off of the wire until only the wire is in the chest. Again, note the markings on the tube and ensure you are still at the appropriate depth in the chest. Finally, take the soft chest tube and feed it onto the end of the wire. Slowly advance the chest tube, until you are within 2 cm of the chest wall. Ensure you are able to grasp the wire coming out of the hub of the chest tube. If you cannot, slowly pull the wire out of the chest, and push the chest tube forward, keeping the distance approximately 2 cm away from the chest at all times. Once wire can be grasped from the hub of the chest tube, stop removing wire from the chest. Grasp and hold the the wire at the chest tube hub steady, and slowly advance the chest tube until it touches the chest wall. The wire should now be sitting out of the hub several centimeters. Keeping the wire, and chest tube hub in one hand, bend the chest tube and wire towards the insertion site. Using two hands (and still holding onto the wire and chest tube hub), push the tip of the chest tube through the chest wall, and into the pleural space. As soon as it begins to easily advance into the chest, straighten out the chest tube and now push the chest tube into the chest while holding the guidewire steady (do not pull it, do not advance it, feed the chest tube off of the guidewire). Once the chest tube is at the desired depth, the chest tube can be held steady and the guidewire removed. Immediately, connect a 3-way stopcock and syringe, to prevent air from entering the chest. Suture in place and apply a dressing to the insertion site. Once in place and secured, empty the fluid or air from the chest by gentle suction.

Thoracostomy Tube Placement (Trochar type)

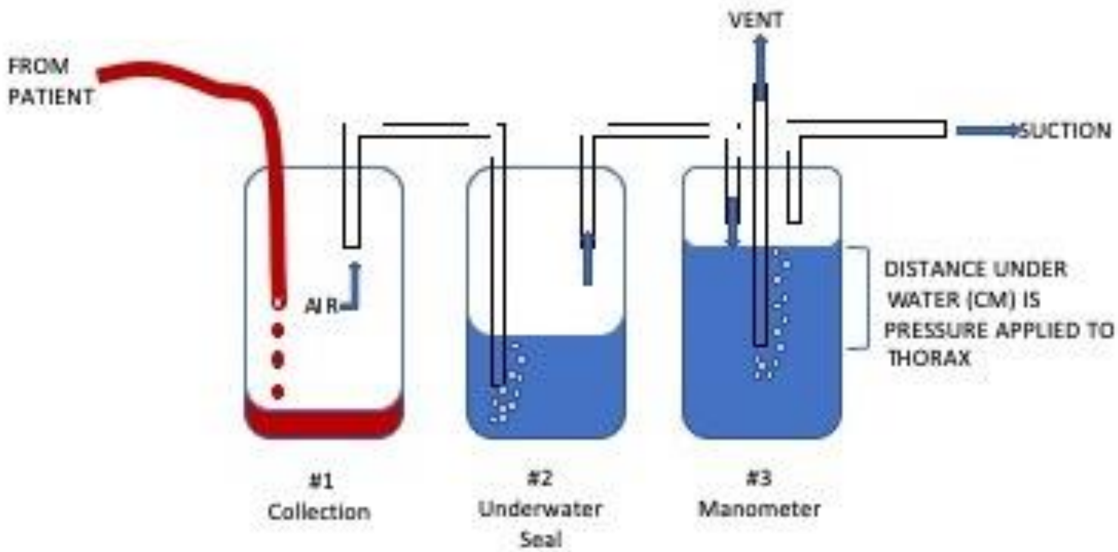
Procedure

- Sedation and local intercostal nerve blocks are used for the critically ill animal.
- A large section of the lateral thorax is surgically prepared.
- The tube chosen should be approximately the same size as the mainstem bronchus (as estimated from a thoracic radiograph). If a trochar type, pull the trochar back so the sharp end is not sticking out.
- The skin is grabbed at the level of shoulder blade and pulled cranially or caudally. (Use the mnemonic fluid-forward, air-back to determine the direction of pull.)
- A stab incision is made (slightly larger than the tube) into the seventh or eighth intercostal space.
- The clinician bluntly dissects through the incision into the thorax with hemostats. The hemostats are held so that the dominant hand is grasping the instrument near the tip of the hemostats to prevent overpenetration.
- Air is allowed to enter the thoracic cavity (to allow some lung deflation).
- The chest tube is placed in a cranioventral direction for fluid and craniodorsally for air. The trochar can be used to give rigidity but should not be pushed from the end (risk of impalement). Rather, work closely to the chest wall, and push the tube gently through the incision that has been made.
- The skin is released to create a subcutaneous tunnel.
- The tube is twisted 180 degrees in each direction to confirm that it is not kinked. Radiographs can be used to confirm proper placement.
- The tube is secured to the periosteum of ribs (using additional local anesthetic).
- A purse-string suture is tied around the tube as it exits the skin to seal the chest.
- The tube is anchored by placing a suture through the skin and around the tube at about the level of the eighth or ninth rib. A Chinese fingertip is placed around the tube to prevent it from slipping out of the chest.
- Triple-antibiotic ointment is placed at the insertion site to create a better seal.
- A wrap is placed around the chest to provide better stability.
- The tube can be connected to a thoracic drain pump for continual suction, or a three-way stopcock can be stabilized to the tube for intermittent suction.
- With these large chest tubes, analgesia is CRITICAL. Ensure that the patient has an excellent analgesic routine, is monitored closely and pain scored, and whenever aspirating from the tube, it is done so very gently.
- Lidocaine is infused into the chest tube (1.5 mg/kg). The chest tube is then flushed with saline (1-3 ml depending on the size of the chest tube). The patient is gently rocked and rolled for a complete coating of the area. Bupivacaine is then infused into the chest tube (1.5 mg/kg) and the chest tube is again flushed with saline. The patient is once more gently rocked and rolled for a complete coating of the area. This treatment can be repeated every 6 hours for pain management. The dose may need to be adjusted in cats if the animal becomes too sedate.
- Heimlich valves can be used for animals with pneumothorax that weigh more than 15 kg.

***** The Trochar Thoracostomy section is from the author's Respiratory Emergency Chapter18 Small Animal Emergency and Critical Care for Veterinary Technicians 3ed, Battaglia and Steele, used with permission.**

Continuous Thoracic Suction

Occasionally, a patient requires very frequent aspirating of the thoracic drain in order to remain comfortable. In most cases, this is because of a significant pneumothorax. With a pneumothorax, it is also possible never to achieve negative pressure during aspiration, and this is a clear sign that the patient may benefit from continuous suction. Continuous Thoracic Suctioning and Chest tube placement should only be performed in hospitals with 24-hour observation and care. Common devices available for veterinary use in North America include Sentinel Seal™, Pleuravac™, and Thoraseal™ to name a few. Each of these compact units is built along the premise of a 2 or 3 bottle apparatus as pictured below:



Bottle # 1 is the collection bottle for liquids, air is moved to Bottle #2, where it bubbles out underwater, preventing any air from going backwards back into the chest in the event of the suction being turned off. Bottle #3 is the manometer, or adjustment for the system. Typical wall suction is set at 160mmHg, which would be far too high a pressure and very uncomfortable for the patient. The vent is used to reduce the negative pressure, typically between 5 and 10 cmH2O.

GASTRIC LAVAGE & ABDOMINOCENTESIS

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Orogastric Intubation (for Gastric Lavage or Decompression)

Gastric lavage (GL) involves orogastric intubation and in veterinary medicine most often performed to remove gastric contents or deflate a distended abdomen as in the case of gastric dilatation and volvulus (GDV) or gastric distention. In human medicine GL has shown no benefit over activated charcoal so many times they no longer perform GL. In veterinary medicine it has perceived to have some benefits with some kinds of toxins. The benefits should outweigh risks. Risks include aspiration and perforation.

The technique for lavage versus decompression is fundamentally the same except that in orogastric intubation for decompression (for GDV) the patient is generally not sedated and in gastric lavage it is sedated. Measure the tube from the nares to the last rib. If performing orogastric intubation manually close the pet's mouth over a large 2", elastikon or vetrap roll. You can also use a bandage roll like vetrap to secure the dog's nose over the roll. If you are sedating for lavage be sure to have all supplies ready and monitor the patient like any other anesthetized patient. Mark the position of the tube. There are various size tubes. Be sure to pick one that is the appropriate size for your patient's esophagus. If the pet starts to regurgitate or vomit, remove the tube immediately and point the head downward to help reduce aspiration of contents. Keep the patient's head in a 90 degree (L shape) normal anatomical position. Lubricate the tube heavily with water soluble (KY Jelly) jelly. Use firm, but gentle pressure to slowly advance the tube. If you feel resistance you should stop. In the case of GDV this could be an indication that you are up against a twist. Continuing to advance may lead to perforation. Apply a gentle twist motion (like a drill) when pushing in to the esophagus. Once in the proper location, place the tube down below the patient in to a bucket. If nothing is flowing, you may need to suction the tube to start the gravity flow. If still nothing is flowing you may have an obstruction in the tube. Instilling 10-60ml of water can be infused in to the tube to flush any obstruction and start the flow of contents. Before removing the tube, kink it off to prevent any remaining contents from dripping down the trachea on removal. In the case of gastric lavage you will want to remove contents and then instill water to flush the stomach. Most references will suggest starting with 5-10 ml/kg of water for each flushing cycle. You can give up to 20-30 ml/kg per cycle. After instilling the water, allow it to drain passively in to the bucket below. Activated charcoal can be administered in to the tube prior to removal.

It is this author's recommendation that when performing gastric lavage you do so with the patient fully anesthetized. Sedation alone can have a lot of risk factors including a patient who is disoriented and thrashes about biting on the tube. Full anesthesia will allow you to intubate with an endotracheal tube, eliminating the risk of placing the orogastric tube down the trachea. With a cuffed ET tube you also minimize the risk of aspiration. The anesthetized patient should have a very good swallowing reflex prior to extubation to reduce risk of vomiting or aspirating down the trachea.

Abdominocentesis

Blind abdominocentesis is more common than ultrasound guided. It is used to detect and collect fluid in the abdomen. Gastric trocarization (Gastrocentesis) can be performed in patients experiencing GDV. A 2013 study showed no statistical difference was found between the gastric trocarization versus orogastric intubation with regards to survival rate or complications. In theory orogastric decompression should result in a greater emptying of the stomach, but a gastrocentesis is less stressful to an awake patient. In both procedures you want to place the patient in left lateral recumbency to avoid puncturing of the spleen. Some references will suggest leaving the patient in left dorsolateral recumbency for gastro or abdominocentesis. Clip and prep the ventral abdomen. Wear sterile gloves.

The main difference between an abdominocentesis and gastocentesis is where the needle/catheter is inserted. For an abdominocentesis, insert a 20-22 gauge needle or over-the-needle catheter cranial or caudal to the umbilicus 1-2cm to the midline. Many references will suggest performing a 4 quadrant technique where you will scrub 2-4" (size of patient dependent) from the umbilicus in a square. Using the umbilical as a center you will think of it as a square quadrant and place needles starting in one of the areas. Remember the point is to detect or collect fluid. Once you have obtained fluid you can stop with the procedure. You don't need to puncture the other quadrants if you get fluid on the first stick. Depending on the goal you can attach an extension line or even an empty fluid bag/line to the end of the needle/catheter to drain the abdomen of fluid or you can attach a syringe and obtain a sample collection. In the case of removing gas from the stomach, use a 16-18g over the needle catheter (or needle) and place it in to the left area where the stomach is. Once you hear gas you can then just keep the pet still so allow for decompression of the stomach.

NASAL FEEDING LINES

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Feeding Tubes

When a feeding tube is placed in a pet it is done so to allow for appropriate nutrition of the pet until they are able to intake the appropriate nutrition orally. In general after a feeding tube has been placed the pet is started off slow (about ¼ maintenance) to see how the pet handles the nutrition being given. If animal shows abdominal discomfort, vomiting or diarrhoea the caloric intake is either discontinued or decreased by a ¼. As the pet is tolerating the feedings, the caloric intake is slowly increased every day until the pet is receiving its required caloric dose. The amount of food required daily is divided in four feedings or is given at a constant rate infusion.

Nasoesophageal (NE) tubes (a feeding tube that ends in the esophagus) can be used for short term nutritional support. It is contraindicated in animals that are still vomiting. Mild sedation may be required in order to place the NE tube.

1) Tilt the animals head up and place 2-4 drops of lidocaine, proparacaine or bupivacaine into the nostril.
2) Measure the feeding tube from the tip of the nose to the fifth intercostal space and mark it with a marker. A 5fr can be placed in most cats and, depending on the size of dog, an 8fr or greater can generally be placed. If the pet is very small a 3.5 fr can be placed, but the solution entering generally needs to be very diluted or it will clog.
3) Lubricate the tip of the catheter with a water-soluble lubricant.
4) Grasp the animal's nose (or head if a cat), keep their head in a normal position and briskly insert the feeding tube.
5) Once the tube is passed to the level of the mark, secure it with suture to the nostril.
6) Check tube position

There are many different techniques on how to fasten a NE tube, but no matter what the technique, the important part is to make sure it is secure (so it cannot slip out) and the patient is as comfortable as possible (so the tube is not lying over their eyes). Tube position can be checked by taking a lateral radiograph or inserting a small amount of water 3-5mls through the tube and observing for a cough reflex. Negative pressure with a 10-20 ml syringe can also be observed if it is in the esophagus, while air will be obtained if it is in the trachea. Most patients will need to wear an Elizabethan-collar to prevent them from pawing at their face which will result in removal of the nasal line.

Nasogastric (NG) tubes (a feeding tube that ends in the stomach) are placed in a similar fashion as a NE tube except that the tube is measured to the 13th rib. Some veterinarians prefer a NG over an NE tube because you are able to aspirate stomach contents out (like excessive stomach acid). However, others worry it can cause an increase in gastric reflux because it interferes with the gastric sphincter. Studies have not proved a benefit of an NG over an NE tube. To place, the same technique used for a NO tube should be tube except the tube should be measured to the last rib. Pushing the nose upward helps moving the tube downward into the ventral meatus. If the pet starts to cough, the tube likely entered the trachea. After placement tube positioning should be checked by using the same technique described above.

NE and NG TUBE CARE

Most commonly a commercially prepared liquid diet is given through a NG or NO tube. The liquid diet is usually given at a constant rate infusion to allow for continuous nutrition until the pet is ready to intake food orally. When the feeding tube is not in use it should be flushed with a little water to ensure that it does not clog. If the tube clogs it is imperative to check to make sure there is not a kink in the line first. If there is no kink and the clog is most likely a result of food that has become lodged then you can try to push warm (not hot) water into the line. A small amount of seltzer water or cranberry juice may help to break up the clog. You can also try to aspirate back on the line to see if adding a "suctioning" pressure may help to free up the clog. Lastly, you can squeeze any visible line with your fingers in an effort to "crush" and break up the clog. Unfortunately if the clog is significant the tube may need to be pulled and replaced.

VENTILATOR SETUP AND NURSING CARE

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Patients that undergo mechanical ventilation present a combination of situations that must be addressed to ensure the best chance for recovery for the patient. Some of these issues arise due to the underlying problem that led to the need for mechanical ventilation (example: cervical myelopathy or heart failure); while others arise from the anesthesia requirements and the prolonged recumbency associated with mechanical ventilation. These patients are the epitome of critical care nursing and require 24/7 dedicated care by specially trained veterinary technicians/nurses and veterinary specialists overseeing their management. Ideally, patients requiring mechanical ventilation will have a 1:1 technician/patient ratio, with other team members available to assist as needed. These are not patients that can be periodically checked on. They always require vigilant monitoring, with special care to ensure that any problem is identified and addressed as early as possible. Communication between team members is vital to ensuring smooth operation and hand over of patients between shifts and during times of concern.

Mechanical ventilation can be categorized based on the anticipated duration that the patient will require assistance. Short term mechanical ventilation is for periods lasting less then twenty-four hours. Long term mechanical ventilation can last days to weeks, to potentially for as long as the patient requires it and is able to be sustained without complications. An important key to successful mechanical ventilation includes training and preparation. This means that all team members who will be responsible for caring for patients undergoing mechanical ventilation have been appropriately trained on how to use and trouble shoot the equipment. While it is not necessary for everyone to be an expert with the ventilator and associated equipment able to solve every conceivable maintenance and troubleshooting error, they should be confident in its operation, able to set it up and trouble shoot basic problems. They should understand its different mode of operations and how to adjust the fraction of inspired oxygen. They should be confident in the operation and troubleshooting of any accessory equipment that is required, such as syringe and infusion pumps, suction devices, warming devices, feeding tubes, various vascular access equipment, multifunction monitors, etc. It is not enough to be able to perform the responsibilities asked of us only if the patient remains stable. The time to train and prepare for problems is BEFORE we are tasked with taking care of a patient. We need to be able to intervene and trouble shoot any equipment errors while maintaining the stability of the patient. Just as it is in CPR and other responsibilities as credentialed veterinary technicians, practice and preparation will improve the outcomes and likelihood for success.

These patients are at an increased risk for ventilator associated pneumonia, along with other systemic infections. It is vital that we take steps to maintain a clean environment. Remove unnecessary equipment and clutter in the area the patient will be hospitalized. Disinfect surfaces and equipment on a regular basis. Hand washing should be employed every time before interacting with your patient. Examination gloves should be worn to reduce the possible transmission of bacteria and other contagions. Sterile gloves and aseptic techniques should be used whenever dealing with the respiratory tract or any procedure where such care is warranted (i.e. urinary catheter placement).

General anesthesia (or deep sedation) is generally required for patients that will undergo mechanical ventilation. The protocol used should be tailored to the needs of the individual patient. There is no such thing as one ideal protocol for mechanical ventilation. We need to ensure we address the underlying issue(s) necessitating mechanical ventilation, as well as comorbidities (both present at the onset of mechanical ventilation, and those that may develop over the duration). In general, total intravenous anesthesia (TIVA) is utilized to maintain the depth of anesthesia desired. Inhalant anesthetics can inhibit the body's compensatory mechanism for pulmonary vasoconstriction during times of hypoxemia, leading to increased V/Q mismatch, and increasing the hypoxemia the patient experiences. This is particularly true for patients undergoing mechanical ventilation with a primary respiratory issue leading to hypoxemia. Prior to induction for the purposes of mechanical ventilation, pre-oxygenation should be attempted for a period of 5 minutes with 100% oxygen. This may not always be possible in critical patients requiring rapid sequence induction to stabilize them but should be attempted whenever practical and safe for the patient. When using TIVA, single or multiple drugs may be utilized to achieve the desired level of anesthesia. Multiple drug choices using a balanced multimodal approach may be preferred to minimize any negative effects on the cardiovascular or other body systems. Patients that have severe systemic illnesses will not likely require as much drugs as a patient that is healthier. Combinations such as a benzodiazepine and an opioid are common, with the addition of ketamine, dexmedetomidine, and/or propofol as needed. All anesthetic agents used should be titrated to the smallest dosages required to achieve the desired effects. Evaluation for the depth of anesthesia should be conducted at least hourly, or at any time that there is a change in the vital parameters monitored that warrant concern. The ability to titrate your anesthetic agents is best achieved with each medication being administered independently of the others. This often leads to the "tree of life" setup with multiple syringe and infusion pumps. Ensure proper labeling of all IV lines and pumps to avoid human error. Make sure to follow any recommendations/restrictions as to the storage of the anesthetic agents. Ask yourself how much drug can be safely drawn up for this patient based on expected usage? Do I need to protect this medication from light? Label all syringes/bags/burettes with the drug(s), diluents, time they were prepared, and any discard deadlines as applicable. The reduction of stimuli can help to reduce the need for anesthetic agents. Place cotton balls in the ears to minimize the perception of sound. Cover their eyes or face to reduce stimulation from light. When palpating or touching the patient, avoid rapid movements or painful stimuli that may rouse the patient.

As previously stated, vigilant monitoring is always required for mechanical ventilation patients. This will include not only equipment monitoring but being hands on with your patient for maximal information and assessment. Multifunction monitors can be convenient to save space and provide information in the same space; however, it is the parameters you are monitoring that are more important than whether you have individual monitors or an all in one. At minimum, one should have in place the ability to continuously monitor ECG, pulse oximetry, capnography, temperature, and either serial or continuous blood pressure. It is helpful to utilize pre-formatted forms specific to mechanical ventilation or critical care patients to ensure the documentation of vitals and allow for a visual trend to be observed upon review. We need to ensure that we are monitoring the patient and not just a specific set of numbers. To do this, I recommend using Kirby's Rule of Twenty as a basis to ensure that all body systems and critical needs are addressed at least once daily. I will review some of the specific issues/body systems that need special attention in mechanical ventilation patients.

The cardiovascular system should be assessed at least every 4 hours with appropriate adjustments made based on the findings. This should include auscultation of the chest and comparing the pulse quality. The ECG should be reviewed, with the potential for multiple leads compared as indicated. Blood pressure may be obtained via Doppler, oscillometric, or direct/invasive methods using an arterial catheter and transducer. Direct blood pressure is the gold standard for care, but may not always be possible, particularly in cases of long-term mechanical ventilation. Doppler or oscillometric blood pressures may be obtained as frequently as needed, but you should balance the stimulation that will occur with the benefit of knowing the results. Time frames of every 5-30 minutes are typically utilized based on the stability of the patient. If a multi-lumen central venous catheter is in place, measurement of central venous pressure may be considered, either continuous (use of a transducer) or every four hours during CV evaluation. Monitor mucus membrane color and capillary refill time, along with hydration status for changes in hemodynamic stability.

The respiratory system should be assessed at least every four hours with appropriate adjustments based on the findings. This should include auscultation of all four quadrants of the thorax. If an arterial catheter is in place, arterial blood gas evaluation may be employed every 4-8 hours as needed. This will help to monitor PaO_2, $PaCO_2$, pH, and enable tweaking of the ventilator to ensure optimal settings. If arterial blood gases are not available, oxygen saturation via pulse oximetry and capnography can be utilized to monitor ventilation and oxygenation status. An endotracheal tube or tracheostomy tube will be in place to facilitate mechanical ventilation. During intubation, a sterile tube should be utilized, along with sterile gloves and technique. Whenever the airway is being examined or treated, non-sterile exam gloves should be worn. Both techniques are important to minimize the possible contamination & introduction of new agents into the respiratory system that could contribute to ventilator associated pneumonia (VAP). Low pressure cuffs should be employed to minimize the occurrence of tracheal necrosis, which has been associated with cuff pressures exceeding 25 cm H_20. Ideally, a commercially available or make your own device that can measure cuff pressure should be utilized to measure and adjust cuff pressure. The decision to move and/or replace the endotracheal (or tracheostomy) tube should be made based on the risk factors for that patient. Both moving the tube and replacing it entirely has been associated with increased risk for VAP in humans. Not moving the tube can lead to tracheal necrosis due to decreased blood flow, while not changing the tube can increase the risk of secretions occluding the tube the longer the tube is in place. Repositioning should be considered every 4 hours if measurement of the cuff pressure is not an option. Replacement of the tube should be considered every 24-48 hours or as needed for patients at an increased risk of occlusion. If utilizing an endotracheal tube, a non-porous material should be used to secure the tube in place (not tie gauze). The use of gauze or other porous materials has been associated with the development/colonization of bacteria as the oral secretions build up/adhere to the gauze. Intravenous extension tubing can be cut down and utilized for securing the tube. The tie should be loosened and moved every four hours to reduce the incidence of ulceration to the lips. Every twenty-four hours, the tubing should be replaced entirely to reduce the potential for a biofilm (bacterial colonization) from forming. Before any movement of the tube for repositioning, replacement of the tube, or replacement of the tie, oral cleaning and suctioning of the mouth and tube should occur to reduce the risk for complications.

Oral care consists of both the examination and cleaning of the oral cavity and should be performed every 4 hours. It allows for early identification of ulcerations, ranula formation, and reflux from the stomach. Cleaning of the oral cavity can decrease the amount of bacteria present in the mouth as well as decrease the overall amount of oral secretions. As swallowing is inhibited in the anesthetized patient, oral secretions can build up. This provides for a growth medium for bacteria and can lead to increased chances for VAP, ranula formation, and ulcers. Documentation and pictures can be utilized to monitor any issues within the oral cavity. Cleaning is usually achieved using a dilute chlorhexidine solution (0.05%). This is used both within the oral cavity, but also on the pulse oximetry probe, mouth gag, and any other associated items within the mouth. You want to suction both the rostral part of the mouth, but also the caudal portion of the oropharynx to reduce any residual chlorhexidine and oral secretions. Be gentle and try not to cause excessive stimulation during cleaning and suctioning, as this can lead to regurgitation and arousal of the patient. When replacing the probe and mouth gags, try to rotate their locations. The tongue should be wrapped in gauze that has been soaked in either saline or a glycerin-based solution. You can alternate between the two if both are available. This helps to prevent drying and ulceration of the tongue. Avoid wrapping the tongue circumferentially, as this can predispose to ranula formation. You may also restrict blood flow if applied tightly. Gently drape the gauze around the tongue instead.

Suctioning of the airway can mean the difference between life and death for your patient in times of crisis. To help prevent getting to the point of airway occlusion and respiratory arrest, suctioning should be performed every 4 hours or as needed. Suctioning the airway and suctioning the oral cavity should be performed as two separate and distinct steps, as sterile technique should be employed for suctioning of the airway. Before suctioning, preoxygenation with 100% oxygen should be performed for at least five minutes. Remember to reset the FiO2 after completion of the suctioning and ensuring the stability of your patient. The suction catheter should be less than 50% of the internal diameter of the tube. It should be soft and flexible and have multiple openings at the distal end to reduce the chance of occlusion. For patients with heavy secretions, you should be prepared to employ multiple suction attempts using a sterile cup or bowl filled with saline to help clear the suction catheter between attempts. Make sure to measure your suction catheter to know how far you can insert it. You do not want to pass your suction tube farther then the distal end of the endotracheal tube, as a cough reflex may result. Other risks if you pass the suction catheter to far include tracheal irritation and bradycardia (vagally induced). Each attempt at suctioning should last no longer then 10-15 seconds, with oxygen supplied in between attempts. You can discontinue your attempts when you no longer see or feel any secretions being aspirated. A new sterile suction catheter and sterile cup/bowl will be required for each separate suctioning event (q4 hours or as needed). The suction tubing and container should be replaced every 24 hours to reduce bacterial colonization. Note that you will need an assistant or technician to work with you during suction events to maintain your sterility. They should wear non-sterile exam gloves and be responsible for the disconnecting/connecting of the breathing circuit. Suctioning does have risks and should always be weighed against the benefits. Pulse oximetry should be monitored continuously during suctioning. Any indication of hypoxemia developing should result in immediate discontinuation of the suctioning and oxygen therapy reinstituted. Other risks include tracheal irritation/inflammation, iatrogenic bacterial translocation/introduction into the lower respiratory tract, bradycardia, hypotension, and collapse of alveoli. A sterile replacement endotracheal tube or tracheostomy tube with full set up, should always be ready and available in case an emergency should arise requiring the replacement of the tube to secure the airway.

Humidification of the airway/breathing circuit is required for patients undergoing long term ventilation due to the drying nature of oxygen therapy. Normally the nose and mouth add humidity to the inspired air; however, we are bypassing those normal mechanisms. If we don't add humidification to our system, we can increase the viscosity of the mucus leading to tube occlusion. The method for humidification will depend on your specific set up/equipment. This may be a heat moisture exchanger or hot water humidifier. Each system has its benefits and drawbacks.

As our patients are recumbent and may be so for a long duration of time, we need to address these conditions as well. Prolonged recumbency can lead to decubital ulcers, tissue necrosis, atelectasis, muscle and ligament contracture, peripheral edema, etc. The platform the patient will reside should be well padded. This may be a table, a gurney, or even the floor for larger patients. Regardless of the location, the area should prevent the limbs from hanging over the sides. The area must be kept clean and free of unnecessary clutter. Passive range of motion should be carried out every four hours. This includes not just the major joints such as the hip and stifle but should continue down the limbs manipulating every joint through the toes. The position of the patient needs to be changed every two to four hours as well. If the patient's condition will tolerate it, the patient should be rotated between lateral recumbency of each side and sternal recumbency. If the patient cannot be placed completely lateral, then the front half should remain in sternal recumbency. The hips and hind legs should be rotated to change the side down. Small adjustments may be made to the front half, so that the pressure points might be shifted. Care must be taken to watch for signs of edema or ulcer formation, particularly around the elbows and chest, which will not be getting a rest during this time.

As with other patients undergoing anesthesia, we need to pay attention to our patients' eyes. Blinking allows for normal tear production to be spread across the cornea. This reduces the friction on the corneal surface and helps to "wash away" any debris and bacteria that may come in contact with the eye. While we may be covering the eyes/face to reduce environmental stimulation, we must not allow this to divert our attention from lubricating the eyes and monitoring them for signs of ulceration and scarring (exposure keratopathy). Every two hours we should rinse the eyes with sterile saline. Check for chemosis, any surface changes (dents) that may indicate an ulcer has formed, conjunctivitis, and vascularization. A fluorescein stain should be utilized once daily for a more thorough evaluation for corneal ulcers. A petrolatum-based lubricant should be applied to the eyes every two hours. These tend to maintain moisture longer then water-based lubricants. If a corneal ulcer is identified, appropriate broad-spectrum antibiotic therapy should be instituted. If an antibiotic ointment is used, then the petrolatum lubricant does not need to be applied at the same treatment time.

As with other recumbent patients, care must be taken to address their urinary system. It will be at the discretion of the attending veterinarian to decide if a urinary catheter should be placed versus intermittent palpation and expression. For larger patients where appropriate bladder palpation may not be achievable, a urinary catheter should be highly considered to minimize the chance of bladder atony. Patients expected to receive mechanical ventilation for more than a day should also be considered for a urinary catheter to minimize the repeated trauma to the bladder wall during manual expression. If manual bladder expression is chosen, the bladder should be palpated every 4-6 hours. Absorbent pads (pee pads) can be used to collect and then be weighed to estimate urine output. Remember that 1 gram of weight is approximately 1 cc of volume. If a urinary catheter is placed, a closed collection system should be employed, and then urine output should be monitored with ins and outs tracked and compared. Urinary catheter care should be performed every 8 hours to reduce the chances of bacterial contamination and urinary tract infections.

Nutrition is not always at the top of the list when discussing emergency and critical care. Yet nutrition is of vital importance in the critical care patient. Nutrition may be administered through enteral and parenteral means. The method(s) chosen will need to be made using a risk/benefit analysis of the patient. Enteral nutrition has the benefit of attempting to maintain the health of the gastrointestinal tract. This can reduce the chance of villous atrophy, GI ulcers, GI bleeding, and translocation of bacteria into systemic circulation. Complications related to enteral nutrition can include increasing gastric residual volume, regurgitation, esophagitis, and aspiration pneumonia. Enteral nutrition can be administered via nasogastric tubes, gastrotomy tubes, and jejunostomy tubes. Esophagostomy and nasal esophageal tubes should be avoided as this may increase the risk of aspiration pneumonia. To help monitor for regurgitation, a food coloring dye that is not normally found in oral secretions, such as purple, may be mixed into the diet. Parenteral nutrition may be utilized solely or in conjunction with enteral feeding to meet the body's nutritional needs. Aseptic technique is required to prevent iatrogenic infections and sepsis. The patient may develop diarrhea during mechanical ventilation. The patient should be kept clean and appropriate sanitary care instituted to prevent inflammation and infections.

Communication and good record keeping cannot be overstated. These patients require intensive nursing care 24/7. The chances for errors can increase with the duration of care as well as the frequency of staff changes. The ability to monitor for trends can help to identify potential complications early and intervene sooner. Communication and preparation will be vital if the patient experiences a crisis, such as a tube occlusion. Mechanical ventilation brings with it the risks of many complications; but it can also mean the only opportunity for successful recovery and discharge from the hospital for some patients. Our ability to provide critical care nursing will directly impact our patients.

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URINARY CATHETERISATION

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Urinary Catheters

Key Points

- Aseptic technique must be used at all times. One complication of urinary catheters is urinary tract infections
- A consistent aseptic technique is required

Function of Urinary Catheter

- To measure urine output
- To assist with nursing care of non-ambulatory or recumbent patients
- To relieve functional or anatomical obstructive disease
- Supportive function for any lower urinary tract surgery
- Temporary or intermittent catheter function
- To allow for urine collection or emptying of bladder
- To perform radiographic contrast procedures
- To determine patency of the urethra or the presence of uroliths (retrohydropropulsion)

Complications of Urinary Catheters

Urinary Tract Infections (UTIs)

- Risk significantly increases the more days the catheter is left in situ
- 20% incident of UTIs following one-time catheterisation in female dogs
- If animals are currently on antibiotic therapy for other disease, then they are more likely to develop a UTI with a resistant bacterium
- Aseptic technique must always be used when handling urinary catheters. This includes wearing gloves while performing any urinary catheter care

Physical Trauma

- Urethra strictures or tears
- Bladder strictures or tears
- Causes
    - Insufficient lubrication
    - Excessive force to introduce the foley
    - Inappropriate location of stylet within the foley
    - Advancing of the foley too far into the bladder
    - Dilation of the balloon when the foley is not placed within the bladder but remains in the urethra, or bursting of the balloon during inflation

Asepsis

- **Wash hands** before attending to the urinary catheter and wear gloves
- **Do not allow urine to flow back into the patient** (turn off the three way tap before you move the patient). Remember to open 3-way again once moved, if using the system where the collection bag is directly attached to the urinary catheter, these bags have a one way valve in them to prevent the urine flowing back into the patient
- **Wipe down** the urinary catheter and flush vulva or prepuce every eight hours with 0.05% chlorhexidine
- Change the **urine bag** every 24 hours
- **Do not allow the giving set end to touch anything** i.e. do not let it sit in the drain or floor while changing the bag. Otherwise the risk of infections is very high
- If the foley is blocked, flush urinary catheter with sterile saline
- **Do not allow the urinary bag and line to drag along the ground**. When moving the patient, hold it up or secure it to the patient
- If there is **faeces/vomit or dirt** on the line, wipe it **clean** with 0.05% chlorhexidine

Catheter Care

- **Wash hands** and wear gloves before maintaining a urinary catheter. Ensure good hygiene and aseptic technique
- **Flush prepuce or vulva every eight hours** and wipe down the urinary catheter **line** with 0.05% chlorhexidine
- Keep the urinary catheter system **closed**
- When moving the patient make sure the 3-way tap is turned off at the patient and then moved. This will prevent urine flowing back into the patient, which can cause infection. Remember to open the 3-way tap again after the patient is moved. Or, if collection bag is connected directly to the catheter there is a one way valve that stops the flow back to the patient
- Change the **urine bag** every 24 hours

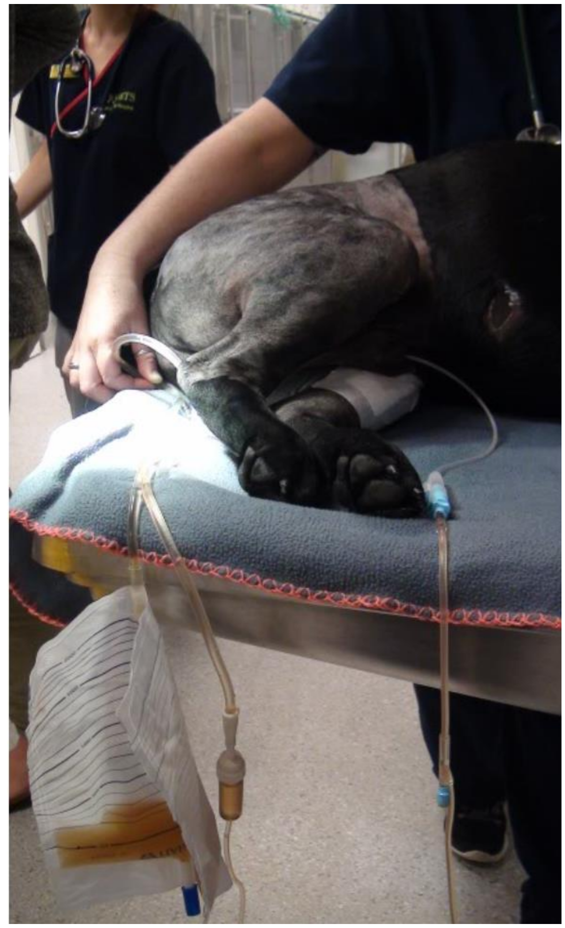
Urine Output (UOP)(monitoring and recording)

- Urine collection system is to be a **closed** system
- UOP should be measured every **four hours or more frequently** for critical cases or patients producing over 1L of urine every four hours
- **Technique**:
    - Collect an empty fluid bag to use as a replacement before any component is disconnected, or a new urinary collection system.
    - Use the clamp to stop the urine flow from the patient (if a fluid bag collection system is used)
    - Disconnect the fluid bag from the giving set (or urinary catheter from the foley) and replace with new fluid bag. When this technique is used, it is EXTREMELY IMPORTANT NOT TO COMPROMISE ASEPSIS! So at no point should the giving set end touch anything, particularly the ground or unsterile areas. Replacing the full bag with a new empty bag is one way to compensate for this
    - Retain the used fluid/ collection bag with urine to measure
    - Unclamp the line and ensure urine continues to flow (if clamp used)
    - If the closed collection system is not being changed to a new bag. Wipe the plug at the bottom of the bag with alcoholic chlorhexidine, pull the plug and empty the urine in the urine jug for measurement. Close the plug and wipe over with alcoholic chlorhexidine. Measure the urine to calculate the urine output
- **Calculating UOP:**
    - The urine collected can be emptied into a measuring cup. A syringe can be used to obtain the mLs.
    - The total volume is then divided by the weight of the animal and then by the number of hours since the bag was last changed:
        - For example: 250mLs/ 25kg dog over four hours = 2.5mLs/kg/hr
        - 250 divided by 25 = 10, 10 divided by 4 = 2.5
    - **Normal** UOPs are:
        - **1-2 mLs/kg/hr** if not on fluids or only on maintenance fluids
        - Generally higher ie >2mLs/kg/hr if on higher fluid rates

What to do with low or abnormal UOP:

- Check fluid rates to ensure that the animal is receiving more than maintenance and/or is actually drinking water
- Check that the 3-way tap is correctly aligned to allow flow (if one used)
- Check that all clamps are off and allowing flow
- Palpate bladder to assess if the bladder is actually empty. If it is empty, then most likely the urinary catheter is functioning correctly. If unsure, ultrasound the bladder
- Flush the urinary catheter to determine if the catheter is patent, remember to remove (if possible) the same amount of fluid or deduct this volume from the total before calculating the UOP
- If any of the problems listed above are noted and action taken then:
    - Check immediately for flow in the line (particularly if bladder size was inappropriate or if flushing improved the flow) to ensure that problem is resolved

- Recheck UOP in 30 minutes to determine if improvement has occurred and that the UOP is in fact accurate
- Record on the patient file



- If no increase in flow or if you are concerned about the animal's status then:
    - Contact the primary case clinician to determine what further action is required. Documenting a low UOP can be extremely important to the outcome of cases and will require communication with primary clinician and usually some further action will to be taken once the underlying medical issue with the patient is identified
    - Record on the patient file

Urine samples

- While measuring the urine, take note for the following:
    - **Smell** – can alter with urinary tract infections. Particularly in long-term foleys such as spinal cases or the critically ill
    - **Changes in colour** of the urine ie blood, cloudy, clots etc
- If changes are noted, **retain a sample** (using as sterile technique as possible) and notify the oclinician. Usually an in-house urine analysis (UA)/ sediment examination will be performed. Once again, treatment plan may alter as a result of these findings.

Female Urinary Catheter Placement (Canine)

Equipment required

- Closed drainage system
- Foley catheter and guide wire (females usually use a 30cm)
- Sterile KY jelly or Xylocaine
- Sterile fluid to inflate the balloon
- Laryngoscope, speculum or otoscope (disinfected)
- Dilute chlorhexidine 0.05%
- Chlorhexidine scrub
- Sterile gloves
- Sterile drape
- Adapters and collection bag

1. **Prepare the patient:** Clip at least 2" area around prepuce or vulva area avoiding clipper burn and skin nicks
2. Scrub with soapy chlorhexidine and water
3. Flush vulva with 0.05% chlorhexidine
4. Using aseptic technique wash hands and apply sterile gloves
5. Place a sterile fenestrated drape over the vulva region, particularly in fluffy patients
6. Placement can be performed by either digital palpation of the urethral opening or direct visualisation with the aid of a light source and speculum
7. Often it is best to palpate or visualise first and then pick up the foley catheter (remember aseptic technique)

8. **Prepare the equipment:** Pick up the foley catheter and insert the stylet into the catheter (lube the stylet before placing into the catheter), dip the end of the catheter in the sterile lubricant
9. **Place the Foley**: Locate the urethral opening (by digital palpation or speculum) and feed the foley into the urethra
10. If you are using digital palpation method then place the tip of your finger just cranial to the urethral opening and apply pressure ventrally. Use your finger to guide the foley towards the urethral opening and to prevent the foley deviating cranially or laterally
11. Slowly advance the catheter. If there is any resistance do not force it.
    If the catheter seems to go in smoothly then bounces after only short advancement you are in the vagina. Try again! Occasionally if the patient is small you may get this bounce once you are in the bladder. Once stylet is removed you will get urine.
12. **Connect the collection bag**: Once the catheter is placed remove the stylet and connect the closed drainage system
13. Inflate the balloon with sterile saline with the volume that is stated on the side of the catheter, do NOT overinflate the balloon

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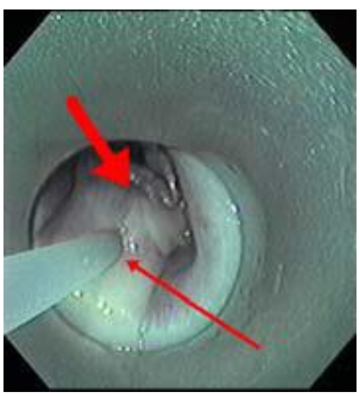
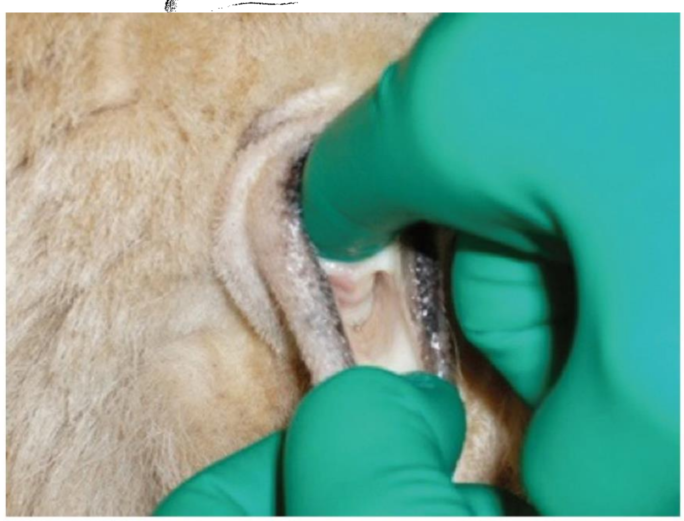
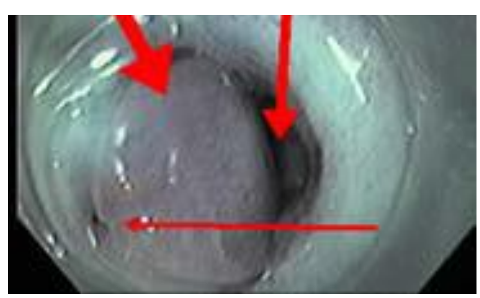




From: Jamie M. Burkitt Creedon and Harold Davis. *Advanced Monitoring and Procedures for Small Animal Emergency and Critical Care.* © 2012 John Wiley & Sons, Inc.

Figure 31.5 The clitoris is in a blind ending pouch, which must be avoided when inserting the finger into the vestibule.



Urinary Catheter – Male Dog

Equipment required:

- Correct size urinary catheter (normally 55cm for male dogs)
- Stylet (if required)
- Sterile gel
- Urinary collection bag system
- Sterile gloves
- Sterile saline to inflate the balloon, either 2 or 5mL
- Drape
- Clippers
- 0.05% chlorhexidine, swabs and syringe (for cleaning the area)
- An assistant to restrain the patient
- Foley adapter *
- 3-tap and injection plug*
- Gravity giving set*
- Urinary collection bag

Size selection

- Normally we use 55 cm for male dogs, for small dogs you may only require a 30cm.
- Large dogs 10Fr.
- Medium dogs 8Fr.
- Small dogs 6Fr.
- Toy dogs 5Fr.
- This is a guide only; try to place the largest size possible for the patient without traumatizing the patient.

Site preparation

- Clip area around prepuce.
- Flush prepuce with 0.05% chlorhexidine.
- Wipe around area with 0.05% chlorhexidine until clean.

Placement and Asepsis

- Ensure aseptic technique.
- Wash hands and dry with lint free towel.
- Glove, with sterile gloves
- With the patient being restrained in lateral recumbency, place a sterile fenestrated drape over the prepuce
- Pick up the foley catheter and insert the stylet into the catheter, dip the end of the catheter in the sterile lube.
- With your assistant extruding the penis, place the lubricated urinary catheter into the tip of the penis (fibrocartilaginous projection of the os penis).
- Slowly advance the catheter, if the catheter gets stuck (as it changes direction at the ischial arch and prostatic section of the urethra), give a gentle push, put do not force.
- Once the catheter is in place, remove the stylet, attach the urinary collection system bag or the foley adaptor, and inflate the balloon.
- If a foley adaptor was used, then attach the 3-way tap to the foley adaptor, and the gravity giving set to the 3-way tap, and a urine collection bag to the gravity set. The urine should flow into the gravity set. Place the injection plug on the free port of the 3-way tap. The original connection on the 3-way tap does not have a complete seal, thus compromising asepsis technique. The 3-way tap should also be turned off to the free port.
- Turn the three way tap off at the patient, move the patient to their cage, and then turn the 3-way tap back on.

Catheter Removal

- The foley will be removed as soon as no longer required. For example, if recumbent animal starts to walk or if UOP monitoring is no longer required.

- The foley balloon must first be deflated. This is done by attaching an empty syringe to the Ffoley balloon outlet and drawing back the sterile saline. Usually this will be 2-5mLs depending on the size of the balloon (it is written on the side of the catheter where the attachments are connected to.
- The foley catheter is then gently withdrawn.

Feline – Urinary Catheters

Indications
- Feline Lower Urinary Tract Disease (FLUTD or blockage)
- Post urethral trauma or surgery to ensure patency
- If UOP monitoring is critical

Equipment
- Slippery Sam urinary catheter (3.5 to 5Fr) and the correct length 8/11/14cm
- Suture material
- Extension 25cm line
- 3-way tap
- Giving set
- Empty IV fluid bag
- Curity tape
- 0.05% chlorhexidine and swabs
- Sterile gloves
- Injection port

Placement of male / female cat catheter
**NB. The technique of placing the cat catheter is described below. Ensure aseptic technique!
- Cats are generally anaesthetised to perform this procedure
- An assistant will be required, to hold the tail +/- prepuce during the procedure and help with equipment
- Clean the prepuce / vulva with 0.05% chlorhexidine prior to catheterisation
- Flush the prepuce with 1mL of 0.05% chlorhexidine solution
- Wash hands and dry with a lint free towel
- Glove, with sterile gloves
- The assistant will aseptically hand the catheter to the gloved person
- The assistant will then extrude the penis or position the vulva so that the person inserting the catheter may visualise the urethral opening
- Insert urinary catheter gently and with the least amount of trauma possible. If any resistance is felt, then discontinue immediately. In the case of blocked cats, small volumes of saline (possibly mixed with sterile lubricant) may be used to help insert the catheter
- Once you have urine flowing out of the catheter, the catheter can be sutured in place
- Attach the 25cm extension line to the end of the urinary catheter for a closed sterile urinary flow. To the opposite end of the extension set attach the male port of the 3-way tap. On the female port of the 3-way attach the sterile giving set and empty fluid bag to produce a closed urinary collection system
- Lastly, with a small amount of curity tape attach the end of the 25cm extension line that is attached to the urinary catheter (the end closest to the 3-way tap) to the tail to ensure that the collection system doesn't pull and drag on the penis / vulva and traumatise it as the cat moves.

Appropriate Care and Monitoring of Catheterised Cat

- Wash hands before maintaining a urinary catheter. Ensure good hygiene and aseptic technique
- Flush prepuce or vulva every eight hours if tolerated, and wipe down the urinary catheter line with 0.05% chlorhexidine
- Keep the urinary catheter system closed
- If any blockages occur, use the 3-way tap to flush sterile saline back into the bladder. With cats, having an assistant is advisable to help restrain the cat while performing this procedure
- When moving the patient make sure the 3-way tap is turned off at the patient first. This will prevent urine flowing back into the patient, which can cause infections. Remember to open the 3-way tap again after the patient is moved
- Change the urine bag every 24 hours

Urinary Catheter Removal

- The catheter will be removed as soon as it is no longer required
- The tape and sutures need to be removed. Depending on the patient, at best this will require a second assistant for physical restraint or may require sedation or other forms of chemical restraint. Check with a veterinarian prior to attempting removal
- The catheter is then gently withdrawn
- Patient should be monitored to ensure normal urination after catheter removal
- The urinary catheter can be discarded

The protocols listed above are reproduced with permission from Veterinary Specialist Services, Brisbane, Queensland, Australia

Suggested readings

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