ANESTHETIC TECHNIQUES FOR THE REPTILIAN AND AVIAN PATIENT

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Birds and reptiles are anesthetized daily for procedures including, but not limited to basic wing and nail trims, venipuncture, radiographs, fracture repair, wound management, soft tissue surgery, etc. Birds and reptiles are not little dogs and cats and those who anesthetize them should be familiar with the normal anatomy and physiology of the specific species they are working with.

Avian Anesthesia

A complete physical examination and at least minimal blood work including a PCV, TP, and blood glucose should be run when possible. Birds can stress easily during restraint and overall handling. Ideally, a pre-medication should be administered prior to any anesthetic induction. Until recently it was believed that birds primarily have kappa pain receptors in the brain. If this is true, full mu opioid drugs may not provide good pain relief for them. If the avian brain primarily has kappa pain receptors, butorphanol is likely the opioid drug of choice used as a pre-medication and an overall analgesic. Recent studies have shown that some avian species have more mu receptors in the brain and actually less kappa than once thought. Full mu opioids will likely provide better analgesia in these species. How do we know what drug will work best? We don't know for most species, but the most current work suggests full mu opioids likely work better in the some hawks and falcons, while kappa agonists work better in psittacines. Opioids are usually given with midazolam IM into the pectoral muscles. Birds and reptiles have a renal portal system. It is believed that some drugs given caudal to the kidneys can potentially be shunted directly to them. If this happens, drugs may not be metabolized correctly.

Most general anesthesia is induced and maintained via an inhalant such as isoflurane or sevoflurane in oxygen. Mask induction is most commonly performed, but chamber or box induction can be used as well. Masks used for dogs and cats can be adapted for use in birds. In some cases, you will need to be inventive when dealing with patients with long or large beaks. Ideally the seal on the mask should be tight so the staff is not breathing is anesthetic gases. I believe mask induction is safer compared to box induction because the patient can be monitored more closely. The anesthetist should ideally listen to the patient using either a stethoscope or Doppler from the time of induction until the patient is extubated. Isoflurane and sevoflurane are potent vasodilators and can have a dose dependent effect on hypotension. These inhalants act rapidly and are only minimally metabolized by the liver. Changes in anesthetic depth can be changed rapidly. As a reminder, in mammals, MAC or Minimal Alveolar Concentration is the concentration of anesthetic that produces anesthesia in 50% of patients that are given a noxious stimulus. MAC is influenced by anesthetic drug protocol, temperature, disease processes, species, stress, and age. Birds do not have an alveolar lung, therefore it is inappropriate to use the term "minimal alveolar concentration" in these species. Instead, we refer to MAC in birds as "minimum anesthetic concentration." MAC in the avian patient is defined as the minimum anesthetic concentration required to keep the patient from purposeful movement to a noxious stimulus. In simple terms, MAC is the lowest percentage of inhalant anesthetic you can administer the patient without it moving in response to being stimulated. Isoflurane and sevoflurane do not provide analgesia therefore an analgesic drug must be used if performing a painful procedure.

Injectable drugs are not generally used due to the lack of studies performed in avian species and the difficulty of pre-placing an IV catheter. Once the bird has entered the appropriate plane of anesthesia, it should be intubated with an uncuffed endotracheal tube (ET tube) and either hand ventilated or placed on a ventilator and provided intermittent positive pressure ventilation (IPPV). The ET tube should be secured by taping it to the lower beak or taping it around the head (similar to what is done in dogs and cats). Most birds do not ventilate well under anesthesia so providing ventilation is a must. A noncuffed endotracheal tube is used because birds have complete tracheal rings and lack elasticity in the trachea like dogs and cats. Using a cuffed ET tube can cause trauma and pressure necrosis to the trachea. This could eventually lead to the need for a tracheal resection or cause death.

The avian trachea is also sensitive to dry air forced through the endotracheal tube. Dry, forced air can cause irritation to the trachea leading to trans-tracheal membrane formation. Trans-tracheal membranes are not common and there is not a lot of information about them. Clinically, if they form, we see them develop about 5 to 10 days after intubation. Most cases are on small birds that were maintained on a pressure driven ventilator, but we have certainly seen them with large birds as well. These membranes are usually the kiss of death. Treatment often requires a tracheal resection and anastomosis. To help prevent trans-tracheal membranes we currently use ventilators with ultra low pressure settings or hand bag the smaller birds.

Adding a Humid-vent® to the endotracheal tube can also help as this disposable device provides humidity to the respiratory tract while the patient is intubated.

As with dogs and cats, anesthetic depth can be monitored using toe pinch, jaw tone, and palpebral reflex. These are all good ways to help monitor the plane of anesthesia. Common monitoring includes HR, RR, venous refill time, MM color, ECG, Doppler with sphygmomanometer and blood pressure cuff, core body temperature, and $ETCO_2$. The temperature probe is placed either "rectally" into the coelom or into the esophagus.

$ETCO_2$ is attached to the endotracheal tube either using a mainstream or sidestream technique. Capnography is a good tool to help assess ventilation in avian patients as this is an indirect measurement of arterial CO_2. $ETCO_2$ correlates well with $PaCO_2$ (direct arterial measurement of CO_2). In mammals, the measurement of $ETCO_2$ can read about 5 to 7mmHg lower than actual $PaCO_2$. In birds this opposite is true. The anatomy and physiology of the avian lung is quite different than that of mammals. Bird lungs create an efficient cross-current exchange system which produces a higher concentration of CO_2 in expired gas compared to the actual arteries. This means that the $ETCO_2$ reading on the capnogram will be higher than the actual $PaCO_2$. In birds, it is estimated that the $ETCO_2$ reading will be about 5mmHg higher than the $PaCO_2$ reading.

In summary, $ETCO_2$ in the avian patient overestimates actual $PaCO_2$ by about 5mmHg which is opposite of what is observed in mammals.

In veterinary medicine, the most common ECG used for monitoring under anesthesia is the 3-lead system. There are various color schemes used for each lead, but most commonly a white lead is used for the right wing web, a black lead is used for the left wing web, and a red lead is used for the left leg or foot. These leads can be placed on the animal using an ECG sticky pad either attached to the foot/wing web or chest/coelomic wall. Alligator clips can also be used, but the teeth should be flattened out when possible. Sometimes these techniques do not work or the patient's skin is too delicate. If this is the case, needles can be pushed through the skin with the alligator clips attached to the needles. I generally find this to be the best option for most avian patients.

Blood pressure is most often taken using an indirect, non-invasive method with a Doppler probe and blood pressure cuff. The Doppler probe is most often secured over the medial metatarsal or cutaneous ulnar (basilic) arteries with the cuff placed proximal to the probe. The probe can be taped into place or two tongue depressors can be taped together to help hold the Doppler over the artery. The width of the cuff should measure about 40% of the circumference of the leg or wing (this is true for any species). There are a variety of very small blood pressure cuffs available on the market today. The most common sizes used for birds are #1 or #2 unless you are dealing with a large bird. One important thing to remember is that improper cuff size can give you erroneous results. If a cuff is too small (this does not happen that often with most exotic animal patients) or above the level of the heart, the blood pressure will be falsely decreased. On the other hand, if the cuff too large or below the level of the heart, the blood pressure readings will be falsely elevated. There are cases where the smallest blood pressure cuff is just too large for the leg. When this happens, I still try and monitor blood pressure knowing that I am mainly looking at trends rather than real numbers. I still feel this is worth doing because I have had cases where the systolic blood pressure readings were consistently around 100 mmHg and then suddenly dropped to 30 mmHg. This told me something changed. In one particular case I asked if there was any bleeding because the blood pressure had dropped drastically and the surgeon told me that the patient had just started to hemorrhage. A severe drop in blood pressure could also indicate the cuff moved or fell off, the patient is too deep, etc.

The Doppler method will only give you systolic blood pressure. To obtain the blood pressure, you must inflate the cuff using a sphygmomanometer. The cuff is inflated until the heart sounds can no longer be heard and then slowly deflated. The first heart sound you hear will correspond to a number on the sphygmomanometer. This number indicates the patient's systolic blood pressure.

Keeping the patient warm while anesthetized is extremely important. The use of a heating pad, forced warm air blanket, heat lamps, etc. can be used. It is important to monitor the patient until it is awake and standing in the cage. Extubation should only take place once the patient's eyes are open, moving the feet and wings, and has good jaw tone.

Catheterization Techniques

As with dogs and cats, an intravenous catheter should be placed when possible. The jugular, medial metatarsal, or ulnar veins can be used for catheter placement. The most common sites are the medial metatarsal and ulnar veins. Depending on the size of the vessel, a 26 gauge to 20 gauge catheter can be used. The medial metatarsal catheter is taped into place in the same manner as a cephalic catheter in the dog or cat. The wing and jugular catheter will need to be sutured into place using a small butterfly piece of tape and a few simple interrupted sutures. If it is not possible to place an IV catheter, an intraosseous catheter should be placed. The IO catheter is generally placed in the ulna or tibiotarsal bone. In birds, the ulna is larger than the radius. IO catheters should never be placed into the humerus or femur. These bones are pneumatic and connect to the respiratory system. Administering fluids into a pneumatic bone can drown the patient. A spinal needle is used to help prevent a bone core from clogging the catheter. The size of the bone will determine the size of the catheter. Generally a 25 gauge to 20 gauge spinal needle is used. These catheters are easy to place but somewhat harder to maintain. Intraosseous catheter placement should be done using aseptic technique. The catheter is sutured in place using a butterfly piece of tape and protected with a bandage.

Fluid therapy should be started while under anesthesia as long as it is appropriate for the patient. The common anesthetic fluid rate for crystalloid fluids is 5mL to 10mL/kg/hour. Colloids can be used when necessary. The dose of whole blood or a synthetic colloid will vary based on the condition of the patient. In general, the rate is started between 2 and 5 ml/kg/hour and is adjusted as needed. Boluses can be given if needed. Because birds are small, a syringe pump should be used to accurately give fluids throughout the procedure.

If a catheter cannot be placed or if the procedure is very short (i.e. radiographs, venipuncture, bandage change, etc.) subcutaneous fluids should be considered. The most common site for subcutaneous fluids is the inguinal area. Others sites include the wing web or over the back. Fluid rates for birds generally range from 50 to 60ml/kg/day.

Post-operative medications should be given for painful surgical or medical procedures. Again, butorphanol or hydromorphone are used to provide various degrees of pain relief in avian patients. When possible, a non-steroidal anti-inflammatory (NSAID) is given to help with pain relief. Meloxicam is the most common NSAID used in birds. Pain medications should be repeated as needed to provide good pain control.

Reptile Anesthesia

Anesthesia can be challenging in reptiles. Pre-medications should ideally be given prior to any anesthetic induction. Common pre-medications include butorphanol, morphine, midazolam, ketamine, telazol, and dexmedetomidine used in various combinations. Pre-medications are given intramuscularly in the front limbs and should be given at least 30 to 60 minutes prior to anesthetic induction. It is important to keep the patient warm enabling them to properly metabolize the drugs.

While the pre-medications are taking effect, you as the anesthetist should prepare for the anesthetic procedure. All instrumentation should be organized and ready to place once the patient has been anesthetized. Common monitoring techniques used for reptiles during anesthesia include HR, RR, MM color, ECG, Doppler, sphygmomanometer, BP cuff, $ETCO_2$, and a temperature probe. The ECG is placed in the same manner as in mammals if you are going to use ECG pads. In some cases, these don't work well due to the scales therefore alligator clips can also be used, but the teeth should be flattened out when possible. Sometimes these techniques do not work or the patient's skin may be too delicate. If this is the case, needles can be pushed through the skin with the alligator clips attached to the needles. I generally find this to be the best option for most reptilian patients.

The Doppler probe is placed in the thoracic inlet for tortoises/turtles or directly over the heart in snakes and lizards. In some turtles and lizards, the Doppler probe can be placed over the carpal artery with a BP cuff placed proximal to the probe. This is one of the only ways to obtain BP in these patients without the use of an oscillometric machine. Oscillometric BP monitoring does not work well in small exotic animals, especially those with a very low or very high heart rate. The $ETCO_2$ is attached to the endotracheal tube. As with birds, a mainstream or sidestream attachment can be used. Due to the potential for shunting, $ETCO_2$ is often quite low in most reptiles. In mammals and birds the normal range is between 35 and 45mmHg. At this time true normals are not known for most reptilian species. Clinically we see the $ETCO_2$ around 20 to 25mmHg. The temperature probe is placed either rectally or into the esophagus. The use of pulse oximetry does not work well in reptiles and birds and is therefore not generally used.

An IV catheter should be placed when possible. The most common site for catheterization in the lizard and snake is the caudal tail vein. Ideally the IV catheter is placed prior to anesthetic induction although this is pretty much impossible in most lizards and snakes. The jugular vein is the most common vessel used in tortoises and turtles. Having an IV catheter in place will provide venous access if an emergency occurs during induction and it also provides easy access for anesthetic induction. If IV access is not possible, an intraosseous catheter can be placed. The technique and equipment used is the same as with birds. It is not possible to place an IO catheter in snakes. In lizards, an IO catheter is often placed in the femur, humerus, or tibial crest. Turtles and tortoises are difficult. IO catheters can be attempted in the bridge of the carapace. IV and IO catheter size will depend on the size of the patient. In general, a 25 to 20g catheter or spinal needle is used.

Once IV access has been obtained (if possible), anesthetic induction can take place. Propofol is the most common injectable anesthetic drug used in reptiles. If a catheter is not possible, propofol can be given using a butterfly catheter or a normal syringe and needle. Propofol is given slowly over a few minutes. Once the patient has been induced, it should be intubated and placed on isoflurane at an appropriate percentage. Sevoflurane can be used, but does not seem to work as well in many reptile species. The patient should be placed on a ventilator or bagged by hand to provide intermittent positive pressure ventilation (IPPV) throughout the procedure since reptiles do not breathe well on their own while under anesthesia. It is very important to keep the patient at an appropriate temperature during the surgical procedure. For most species of reptiles, core body temperature should be kept between about 85 and 90 degrees. If the patient is kept too cold, the drugs will take hours to metabolize and the patient will have a prolonged recovery time. Lactated Ringer's Solution is commonly used at a rate of 5mL/kg/hour.

After the procedure is completed, the isoflurane should be turned off and the patient should be taken off of pure oxygen. The reptile's respiratory drive to take a breath is more oxygen driven rather than carbon dioxide driven as in mammals and birds. If kept on pure oxygen, the reptile patient has a lesser drive to take a breath on its own and recovery can be very prolonged. An ambu bag should be attached to the endotracheal tube and the patient should be given a breath about 4 to 6 times per minute until the patient is awake and can be extubated. During this time the heart rate is monitored using a Doppler. The patient should only be extubated when it has some jaw tone, muscle tone in the limbs, palpebral reflex, and breathing well on its own.

Post-op medications should be given at the conclusion of the surgical procedure if necessary. The most common post-op drugs include butorphanol, morphine, meloxicam, and tramadol.

Suggested Reading

Ballard, B. M. and Cheek, R. (Eds.) (2016). Exotic Animal Medicine for the Veterinary Technician, 3rd ed. Wiley and Sons.

Bryant, S. (2010). Anesthesia for Veterinary Technicians. Wiley and Sons.

Longley, L. A. (2008). Anaesthesia of Exotic Pets. Saunders Elsevier.

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