Approaches to Avian Anaesthesia

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Undoubtedly, avian patients under anaesthesia are far more 'exciting' to monitor when compared to anaesthetised cats and dogs. This, unfortunately, means that anaesthetised birds require constant monitoring and the anaesthetist must pay attention to subtle changes that may determine the patient's outcome for a given procedure. This paper will discuss avian anaesthesia and pain management.

BASIC AVIAN RESPIRATORY ANATOMY

The avian respiratory system has several unique features when compared to mammals. Because of a bird's respiratory anatomy, evaluating and maintaining avian patients under anaesthesia is quite different than with anaesthetised mammals. The following is a simplification of avian respiratory anatomy and the potential clinical significance of selected anatomical features is discussed. For more detailed information, the reader is encouraged to review the cited reviews. [1, 2, 3, 4, 5, 6]

A bird's larynx houses the visible glottis and is connected to a distant syrinx, the avian 'voice box', by a variably elongated trachea. Most birds possess complete cartilaginous tracheal rings. In some penguins, mallards and petrels, a septum divides the cranial trachea into two tubes. [7] Male Anseriformes have a bulbous ossified intrathoracic tracheal expansion. Emus and ruddy ducks have an inflatable sac-like tracheal diverticulum. [7] At least 55 avian species possess tracheal elongations that vary from simple loops to complex convolutions. [1] The syrinx, divides into two primary bronchi that enter the lungs.

Clinical Significance: Complete tracheal rings make the avian tracheal lumen less flexible and more prone to damage from an over inflated endotracheal tube cuff. [5]

Avian lungs expand and contract minimally with inhalation and exhalation. Most birds have 9 air sacs. The paired cranial thoracic, caudal thoracic and abdominal air sacs and single clavicular air sac connect with the lungs and are found within the coelomic cavity. The paired cervicocephalic air sacs are subcutaneous and connect with the infraorbital sinuses. The majority of inhaled air passes first into the caudal thoracic and abdominal air sacs, then on exhalation passes through the lungs. On inhalation it moves into the cranial thoracic air sacs and to some extent into the cavicular air sac and on the second exhalation re-enters the trachea and is expelled.[5] Air sacs are poorly vascularised and account for less than 5% of respiratory system gas exchange.[7] Since avian lungs are fixed, there is minimal expansion of pulmonary tissue during inspiration and no resultant increased surface area for oxygen exchange, as occurs in mammals. Instead, oxygen exchange occurs as air moves through a bird's lungs. The difference is that in mammals, oxygen is exchanged primarily when the lung is inflated and the surface area of the alveoli increases. In birds, oxygen is exchanged only as air moves through the air capillaries.[2]

Clinical Significance: Birds exchange oxygen only when there is a constant flow of air. For this reason, apnoea in birds can result in death more quickly than in mammals.[2]

Birds lack a diaphragm and rely on movement of the ribs and sternum for ventilation.[6] Anything that decreases the size of the air sac, including ascites, enlarged organs, internal masses and outside pressure (rough handling or restraint devices) can reduce the bird's respiratory efficiency. Also, diseases that cause severe thickening to air sacs or impede air flow into or out of them, can impair normal respiration. Dorsal recumbency, by itself, can decrease minute volume in healthy anaesthetisd birds by up to 60%.[8]

Clinical Significance: When handling birds, devices or restraint techniques that prevent normal sternal movement should be avoided.[5, 6] Diseases that directly or indirectly affect the air sacs and lungs can impair respiration, making anaesthesia and general handling more risky.

As is true with all smaller animals, birds have a high surface to body volume ratio and are predisposed to heat loss when not active. When birds are debilitated and/or under anaesthesia, metabolic activities do not adequately support normal body temperature and supplemental heat should be provided. Heat loss is directly proportional to the size of the patient (the smaller the bird, the faster the heat loss). Heat loss can occur through evaporation and is increased by non-humidified, cool anesthetic gases.[9] A common sign of hypothermia is decreased respiration, which can result in decreased oxygen and carbon dioxide exchange. Appropriately providing radiant (overhead heat lamp) warmth is an effective means of maintaining body temperature in anaesthetised birds. Radiant heat appears to be superior to underlying heating pads as a means of supporting anaesthetised birds.[3] The heating lamp should be adjusted so that the temperature on the bird is approximately the induction temperature of the bird. Although more expensive and somewhat awkward, heated air units can also effectively maintain body temperature in anaesthetised birds if they are applied immediately after induction.[9]

Clinical Significance: Unless suffering from hyperthermia and possibly head trauma or obesity, all sick and anaesthetised birds should receive supplemental heat.[6, 10] Over head heating lamps are practical and effective for use during avian anaesthesia to maintain patient body heat.

Dehydration and rarely hypoglycaemia can be noted in both anaesthetised and debilitated avian patients. Anaesthesia combined with blood loss can lead to varying degrees of hypovolaemia (if not already present) and decreased vascular perfusion.[3] Intraosseous (IO), subcutaneous (SQ) and/or intravenous (IV) fluids warmed to ~ 39ºC (100.4-102.2º F) should be considered in all anaesthetised and debilitated patients. Even a low level of supplemental fluids may be beneficial in healthy avian patients undergoing non-invasive anaesthetic procedures.[3] Shock doses of fluids can be given at 30-60 ml/kg/hr safely in most birds.[11] However, balanced electrolyte solutions are more commonly given at 5-10 mls/kg/hr IV or IO to birds under anaesthesia.

Although frequently discussed as a concern, hypoglycaemia has not been shown to occur in experimentally anaesthetised birds.[11] In fact, 5% dextrose given IV in birds, may be inappropriate and can cause significant electrolyte imbalances.[3] Balanced electrolyte solutions, such as Lactated Ringer's Solution (LRS) and Normosol, are therefore recommended during anaesthetic procedures. [3] Hypoglycaemia may be prevented by limiting fasting in most birds to no more than a couple hours prior to surgery.[2, 6] If the avian patient is stable, blood glucose should be determined before adding dextrose to fluids.

Clinical Significance: Supplemental, balanced electrolyte solutions should be considered in all anaesthetised and debilitated avian patients. Dextrose added to fluids should be used judiciously in sick and anaesthetised birds. For most procedures, birds should be fasted only 1-3 hours prior to anaesthesia.[6]

INDUCING ANAESTHESIA

Unless an emergency procedure is required, avian patients should be fasted and in stable condition prior to anaesthesia. A filled crop should be emptied prior to surgery. Regurgitation and aspiration during anaesthesia occasionally occur when food is present in the crop and regurgitation is more likely to occur in birds with proventricular dilitation disease, intestinal foreign bodies and chronically distended crop and in young patients and birds on a liquid/semi-soft diet (lories, lorikeets). If upper gastrointestinal surgery is planned, a 4- 6 hour fast may be more appropriate to better empty the crop.[6] In the author's experience, larger adult birds (> 550 g) undergoing proventricular/ventricular surgery (foreign body retrieval, biopsy, endoscopy) do well under anaesthesia when fasted overnight. This prolonged fast in adult birds seems well tolerated and surgically allows for a 'cleaner' gastro-intestinal tract.

Isoflourane was the anaesthetic of choice for avian patients however even with the higher price, sevoflourane is becoming more popular.[2, 5, 6, 7, 12, 13] Newer halogenated gas anaesthetics, such as sevoflourane and desflourane, offer faster induction and recovery in studied patients compared to isoflourane.[4,12] It should be noted that isoflourane causes significant hypoventilation in birds and is more likely to cause respiratory depression than in birds as compared to mammals.[14] Halothane is no longer considered safe and reliable for avian anaesthesia.[7] Although many injectable anaesthetics can be safely used birds, isoflourane or sevoflourane (gas) anaesthesia is recommended, even for short procedures.[2]

Prior to gas induction, butorphanol should be considered. Butorphanol has been shown to lower the minimum anaesthetic concentration (or MAC) in some parrot species but also lowers the heart rate, tidal volume and expiratory/inspiratory times.[15] As discussed below, intermittent positive pressure ventilation should be considered throughout avian anaesthesia to help counteract decreased respiration, tidal volume and ultimately patient oxygenation.

Birds are induced with 3-5% isoflourane or 5-8% sevoflourane via a mask using a non-rebreathing (Bain) system.[2, 6] Patient size permitting, the bird should be intubated with a small uncuffed (Cole) or cuffed (non-inflated) endotracheal tube once anaesthetised. The endotracheal tube cuff can be carefully inflated to 10-15 cm H_2O (just enough to prevent leakage), if necessary.[6] Cuffed endotracheal tubes may cause trauma that ultimately results in tracheal stenosis.

Due to the small tracheal lumen, mucus can easily plug the end of the endotracheal tube.[5] Avian patients rarely develop tracheal obstructive plugs when maintained with a mask, but are susceptible to aspiration when not intubated. Although atropine and glycopyrrolate can increase heart rate, it is said to increase the viscosity of avian respiratory secretions potentially making tracheal plugs worse and is therefore, not commonly used in birds under anaesthesia.[5, 6, 16]

Waterfowl, and possibly other birds, have a 'dive response' that can prevent normal respiration when the beak is closed.[5] When diving, the beak is closed and the bird stops breathing via stimulation of (trigeminal) nerve receptors.[5] Also as a result of the dive response, the heart rate can decrease up to 50% (in mallards).[8] Administration of 100% oxygen has been shown to abolish the dive response in dabbling ducks.[8] This is noted clinically when inducing waterfowl with a tight mask and when a bird is anaesthetised with an endotracheal tube taped around and closing the beak. When anesthetizing waterfowl, use a large face mask that permits opening of the patient's beak and tie the endotracheal tube (if used) around the back of the bird's head or to the lower beak only.

Some birds possess a *crista ventralis* that may interfere with intubation. This horn like projection can be found in the glottis lumen and is reported in *Anas* (mallards), *Apteryx* (kiwi), Spheniscidae (penguins), *Gallus* (fowl) and *Corvus* (crows) species.[8, 17] If the *crista ventralis* is present, than either a mask or a smaller endotracheal tube can be used.

A properly fitted face mask can be used on smaller birds to control respiration and anaesthetic delivery. The body of the face mask should allow the full movement of the patient's beak in case open-mouth breathing is necessary (as with plugged nares). By taping part of a rubber glove with a central hole to the open side of the mask, a smaller opening is created. The bird's head is fitted through the smaller opening creating a tighter seal. The seal should be tight enough to permit assisted ventilation without the use of an endotracheal tube and is best with small birds that cannot be intubated.

Air Sac Breathing Tube

If the oral cavity or trachea is occluded preventing proper induction or maintenance of anaesthesia or the air sacs need to be medicated, an air sac tube can be used.[5, 6, 18, 19] Patients with oral masses or tracheal obstructions should be mask- (if possible) or box-induced. Anaesthesia delivered via air sac breathing tubes has been successfully used in patients as small as zebra finches.[19]

When anaesthetised, place the patient in lateral recumbency and quickly, surgically prepare the paralumbar fossa (just behind the last rib). A small skin incision is made over the paralumbar fossa (same site as for surgical sexing) exposing a relatively thin layer of lateral abdominal wall muscles. Use right angle forceps to 'punch' through the muscle layer and into the underlying air sac. Place a sterilised endotracheal, red rubber feeding or other tube into the air sac (either caudal thoracic or abdominal air sac, depending on the bird species and placement) and suture (the tube) to the skin. When properly placed, one can hold a down feather over the tube opening to watch for air movement or place a glass slide over the end of the tube and watch for condensation.[19] The tube diameter should be approximately the same size as the patient's tracheal lumen. Due to progressive microorganism infection and air sacculitis, air sac breathing tubes should not be left in avian patients more than 5 days.[20] In addition to infection, air sac breathing tubes can result in coelomic organ damage, life threatening blood loss (from vessel laceration/trauma), air sac damage and subcutaneous emphysema (which is usually self-limiting).[19]

In studied sulphur-crested cockatoos (*Cacatua galerita*), delivering isoflourane and oxygen via caudal thoracic air sac intubation provided a reliable method of maintaining anaesthesia and resulted in minimal alteration in respiratory function similar to endotracheal tube administration. In the same study, clavicular air sac intubation did not provide adequate ventilation or maintenance of anaesthesia.[21]

Injectable Anaesthetics

While gas anaesthetics offer the best advantages to anesthetizing birds, injectable agents have some notable uses. In field conditions where gas anaesthesia is not available, injectable drugs (such as ketamine and xylazine) are useful. Xylazine's use may cause respiratory depression, hypoxaemia, hypercapnoea, exicitment and convulsions in birds. [7] One advantage of the $alpha_2$-adrenergic agonists (xylazine, medetomidine, etc) is that alpha-adrenergic reversal agents (yohimbine or atimpamazole) can be used. Also, heavy sedation with tranquilizers such as diazepam and midazolam can be used to provide muscle relaxation and reduce stress in fractious species. It has been noted that midazolam given to geese, raptors and pigeons caused a (potentially undesirable) sedative effect that lasted for several hours after anaesthesia.[7] The author has used diazepam combined with local lidocaine anaesthesia and supplemental oxygen to successfully perform minor surgeries in high-risk avian patients that were not deemed safe for general anaesthesia.

Both ketamine (a cyclohexamine) and propofol (substituted phenol derivative) can be used as general anaesthetics for minor and short procedures in birds. Ketamine produces a state of catalepsy and is often used with diazepam or xylazine to improve patient muscle relaxation and analgesia.[7] Propofol is giving IV and is used for its rapid onset of anaesthesia and patient recovery. One disadvantage is propfol's ability to cause dose-dependent respiratory and cardiovascular depression.[7]

MAINTAINING AND MONITORING BIRDS UNDER ANAESTHESIA

Birds are best maintained at 1.5- 2.5% isoflourane or 2-4% sevoflourane, but this depends on patient condition, species variation and the procedure. When using a non-rebreathing system in avian patients, gas flow rates should be 660 ml/kg/min.[2] Another formula for oxygen flow is 150-200 ml/kg/min.[7] For smaller patients, rates as high as 1-3 L/min are typically used.[2] This applies for mask use, endotracheal and air sac intubation.

Avian patients are positioned as needed for the given procedure, but are generally in dorsal or lateral recumbency. Unless absolutely necessary for the given procedure, birds should not be placed in dorsoventral (DV) position while under anaesthesia.[16] During anaesthesia, the patient's head should be elevated to prevent reflux of crop contents into the mouth and the potential for aspiration of them. Supplemental heat, ideally provided by a radiant source, is provided to maintain patient warmth. If needed and not already present, IV or IO catheters are placed for fluid support. Balanced fluids (Normosol or LRS) are given at 10 ml/ kg /hr for the first 2 hours of anaesthesia.[6] After 2 hours, reduce the fluid rate to 5 ml /kg /hr in anaesthetised birds.[6] Additional fluids or blood transfusions may be indicated in the case of significant hemorrhage.

As time is an important factor in avian anaesthesia, quickly prepare the patient for surgery. Pluck and aseptically prepare the surgical site, as for any other animal undergoing surgery. Unless used sparingly, alcohol preps may inappropriately cool and predispose small avian patients to hypothermia.[2] Clear surgical drapes (VSP Veterinary Transparent Surgical Drapes[a]) should be used to allow visualization of the underlying bird.[16]

All monitoring devices, such as electrocardiogram (ECG) leads, doppler and thermometer probes, should be placed quickly just prior to, or during surgical preparation. ECG leads are applied in similar locations as those of small mammals (wings replace forelegs).[6] Due to potential trauma from alligator clips, attach the ECG leads to small (27 to 25 g) needles or sterile wire suture. Next, either loop or penetrate the wire/needle through a thin section of skin on the wing or leg. In lead 2, the RS segment of an ECG reading is normally deflected downward in birds.[2, 6] As a result, normal avian electrocardiograms must be interpreted differently than with mammals.

Doppler units are best placed over one of the visible vessels (cutaneous ulnar/wing vein and ulnar artery) crossing over the medial elbow region to give an audible sound with each heart beat. Small blood pressure cuffs can be placed over the humerus or femur and used in conjunction with a distally placed doppler probe to collect blood pressure readings. If blood pressure drops below90 mmHg, give hetastarch at 5 ml/kg IV or IO until blood pressure is greater than 90 mmHg.[22] Usually only 1 or two hetastarch doses are required.[22]

Pulse oximeters, which measure blood oxygenation, are not currently calibrated for birds and the readings may be inaccurate. In one study, the authors concluded that the current 'pulse oximetry is unsatisfactory for routine anaesthetic monitoring in birds.[23] However, pulse oximeters may provide some useful information as to trends in blood oxygenation in avian patients. Pulse oximeter probes have been used orally and cloacally in birds.[6]

With large birds, blood gas samples may also be evaluated.[6] Some devices (usually attached to the endotracheal tube or elsewhere along the anaesthetic system) are capable of measuring end-tidal CO_2 (capnography) and tidal volume. Measuring end-tidal CO_2 in mechanically ventilated birds is the only means to assess acid-base balance even when respiration appears normal.[14] While one study suggested that capnography can effectively be used to monitor arterial CO_2 in African grey parrots (*Psittacus erithacus*), the end tidal carbon dioxide reading consistently overestimated arterial CO_2 by 5 mmHg.[7]

Body temperature should also be maintained throughout surgery. Warm water blankets, forced air warming systems and radiant heat lamps can all be used on avian patients. Long flexible thermister probes are available and can be inserted into the esophagus to the level of the heart.[7] Cloacal temperature can also be monitored but is dependent upon body position and cloacal activity throughout the procedure.[7]

Once anaesthetised, several patient factors should be evaluated to help determine depth of anaesthesia. Patient respiration is very important to monitor and is discussed in depth below. The pedal (foot withdrawal to pinch), palpebral (eyelid closure induced by ocular medial canthus or cere stimulation) and corneal (third eyelid movement in response to corneal manipulation) reflexes can all be monitored in birds. The palpebral reflex is present only in lightly anaesthetised birds.[2] The pedal and corneal reflexes will sometimes remain as slow movements, even in a surgical plane of anaesthesia.[2] With deep anaesthesia, breathing becomes slow and shallow and all reflexes are gone.[6]

Neuromuscular Blockade

There are very few reports of neuromuscular blocking agents being used in birds. The most likely use would be with in-depth eye surgery. One report describes a mallard (*Anas platyrhynchos*) being surgically treated for bilateral cataracts.[8] The ischiatic nerve was stimulated with a train of four (TOF) pattern that was used to evaluated the degree of neuromuscular blockade. Four electrical stimulations incite four equal strength twitch responses in the associated muscle (fibularis longus in this case) in normal animals. As neuromuscular blockade takes over, the fourth twitch diminishes first followed by the third, second and first twitches. Once 70-75% of the neuromuscular junction receptors are blocked, twitch responses begin to diminish. As the neuromuscular blockade wears off, the twitch responses return in opposite order (the first twitch returns first).[8]

In the mallard described above, cis-atracurium (a short acting, nondepolarizing neuromuscular blocking agent) was given at 0.25 mg/kg IV and monitored until the TOF indicated effective neuromuscular blockade. The mallard was mechanically ventilated until breathing unaided which occurred after 90 minutes. Prior to giving a reversal agent in animals receiving non-depolarizing neuromuscular blocking agents, some recovery of the twitch response should be noted. Edrophonium (0.5 mg/kg IV) was given and normal twitch responses to TOF returned within 5 minutes. Atropine (0.04 mg/kg IV) was giving prior to the edrophonium in effort to counter potential adverse parasympathetic effects.[8]

Respiratory Rate and Character are the Most Reliable Indicators of Depth of Anaesthesia in Avian Patients.[2] Anaesthetised birds should ideally take normal sized breaths at a regular rate. Evaluating respiratory frequency alone can be misleading.[5] The ‘normal’ respiratory rate for an awake resting bird can be calculated at $17.2 \times M_b^{-0.31}$ (M_b is body weight in kg).[24] With this formula, a 100 g, 500 g and 1,000g bird should take 35, 21 and 17 respirations per minute respectively.

There are no set guidelines for normal respiratory rates in anaesthetised birds as there is much individual variation but avian patients should breathe at least once every 2 to 7 seconds. In one report, a mallard given neuromuscular blockade for cataract surgery was adequately maintained with intermittent positive pressure ventilation at 12 breaths per minute.[8] Increased and deep respiration may indicate that the patient is 'light'. In birds, as in and other animals, respiration becomes more shallow and slower as anaesthesia deepens. Heart rate and quality should also be monitored. Decreases or changes in heart rate are often mirrored and preceded by changes in respiration. Phalen notes that a significant (20%) decrease in heart or respiratory rate in anaesthetised birds strongly suggests that cardiac arrest will soon follow and intervention is needed immediately.[11]

Abnormal Breathing Patterns Should Be Identified and Resolved as Soon as Possible. Pathologic conditions, such as pain (surgical manipulation), lung haemorrhage and overheating can induce rapid breathing (**tachypnoea**). With painful conditions, tachypnoea usually resolves by inducing a deeper plane of anaesthesia. Tachypnoea associated with blood filled lungs (hemorrhage) responds poorly to increasing the anaesthetic dose. If lung haemorrhage is suspected or identified (visualised via endoscopy or laparoscopy), anaesthesia should be stopped or continued only with great caution. In birds, lung haemorrhage can be fatal, especially if progressive, its cause is poorly understood and no treatments are currently described. Increased respiration due to overheating is most commonly noted when the avian patient is 'light' or upon recovery, but sometimes, hyperthermia contributes to mild tachypnoea during deeper anaesthetic planes.

Dyspnoea, or difficult and laboured breathing, often indicates poor ventilation. Tachypnoea and dyspnoea can be very similar, are often difficult to distinguish and share some common causes. Underlying diseases (heart, lung and air sac disorders), as opposed to human induced overheating and pain, are more frequently associated with dyspnoea in birds. Dyspnoea may occur due to an obstruction (such as a mucus plug) in the trachea or endotracheal tube, poor cardiovascular perfusion (heart disease), fluid or blood filled lungs (pulmonary oedema, inflammation and haemorrhage) and decreased or diseased air sac space resulting from improper restraint/positioning, ascites, bleeding, organomegaly, other abdominal masses and airsacculitis. Correction of dyspnoea is aimed at identifying and addressing the underlying cause.

Apnoea, or cessation of breathing, is common in anaesthetised birds and can be associated with multiple conditions. If apnoeic birds are not restored (artificially or naturally) to breathing, death will quickly follow. Apnoea is commonly due to poor ventilation, taping the beak closed (diving birds), excessive anaesthetic and hypothermia, but may also be associated with hypoglycaemia, hypovolaemia (blood or other fluid loss) and other underlying metabolic and systemic disturbances. Apnoea is frequently preceded by a decreased respiration rate. Identifying and correcting the causes of decreased respiration rates will help the monitoring nurse prevent apnoea or respond more quickly, should it (apnoea) occur.

When a bird's respiration begins to drop, first evaluate the patient's anaesthetic dose, surgical conditions (disrupting air sacs/lungs, and excessive blood loss) and monitoring tools (body temperature and heart rate). Correct obvious deficits as best as possible by lowering anaesthetic dose, providing fluid support and maintaining patient body heat, as needed. If the respiratory rate continues to decline, auscult the lungs and trachea and determine if the passages are clear. Tracheal mucus plugs will often produce a 'gurgling' sound that may only be heard with the aid of a stethoscope. Lung hemorrhage and edema may also result in abnormal respiratory sounds and should be considered as well. Remove the endotracheal tube (if present) and check for blood or mucus plugs that may be contributing to poor ventilation. In general, endotracheal tubes may need to be replaced every 30 minutes to decrease plug formation. Air sac tubes are an exception, as they rarely occlude with mucus and can be left in for days without problems. However, air sac tubes can become plugged with blood, other fluids and organ tissue and should be checked for patency as needed. The surgeon should be notified when the patient is becoming apnoeic so he/she can identify and resolve any surgical conditions that may be interfering with respiration.

If respiration decreases to the point of apnoea, several steps should be taken to restore breathing. Quickly evaluate the patient as discussed in the above paragraph. If breathing cannot be restored, turn off all anaesthetic gas, maintain oxygen flow and manually ventilate the patient. When manually ventilating birds, provide enough pressure (8- 12 cm H_2O) to create normal inspiratory depth and 10-12 respirations per minute.[6] Some feel that artificial ventilation should be routinely provided to all anaesthetised birds.[5, 6, 11] To prevent volume trauma to the air sacs when providing positive pressure ventilation, limit the pressures to 15-20 cm H_2O.[7]

In birds, decreased ventilation results in increased $PaCO_2$ which directly affects pH and acid-base balance. The expected response in an awake, healthy bird is to increase ventilation, which decreases $PaCO_2$ and increases pH. Anaesthetised birds may not properly ventilate themselves and often require artificial ventilation. In ducks, the common parameters such as respiratory frequency and tidal volume alone cannot be used to monitor acid-base balance. Studies in African grey parrots have clearly shown that intermittent positive pressure ventilation will decrease $PaCO_2$, which subsequently affects pH and acid-base balance.[14] Even birds that are self-ventilating while under anaesthesia should be assisted with 4-6 respirations per minute.

The sternum may be moved ventrally and dorsally to provide ventilation if an endotracheal tube or sealed face mask is not being used.[6] If the air sacs have been exposed as part of surgery, the surgeon may need to temporarily 'close' the surgical site with a finger to help permit more normal ventilation. If the patient is apnoeic, the surgeon must attempt to limit overall surgical time while the attending nurse increases the oxygen flow rate and continues assisted ventilation. Sometimes continued tissue manipulation will help the patient respond and begin breathing. Patients that become apnoeic and are restored should be maintained on a very light plane of anaesthesia for the remainder of the procedure.

Cardiac Arrest Often Follows Prolonged Apnoea or Dyspnoea. Maintaining normal fluid volume, proper oxygenation, ventilation and monitoring, minimal use of cardiodepressive drugs and appropriate anaesthetic depth are all needed to help prevent cardiac arrest. Certainly, some underlying conditions may adversely affect the heart and may be precipitated by anaesthesia. If cardiac arrest occurs, begin cardiopulmonary resuscitation. If possible, place an endotracheal tube (if not already present) and restore ventilation. Doxapram, a positive inotrope, can be given (to help stimulate respiration.[7] Also consider hetastarch at 5 ml/kg IV or IO to increase blood pressure and improve organ perfusion. Manipulate the sternum, as described above, to assist circulation and ventilation.[6] Due to the anatomy of the bird, direct cardiac massage is very difficult. IV, intratracheal or intracardiac epinephrine (0.5- 1.0 ml/ kg of 1:1,000) can be tried to stimulate the heart.[10] Unfortunately, birds are very difficult to resuscitate once cardiac arrest occurs.[6]

Due to the Potential Complexities with Monitoring Anaesthetised Birds, it is Best to Have One Nurse Assigned to Anaesthesia for the Duration of the Procedure. Avian patients require constant monitoring under anaesthesia. As a result, a nurse should always be monitoring anaesthetised avian patients. Assistant nurses are often needed to help gather supplies, adjust and troubleshoot equipment and aid the attending veterinarian or nurse anaesthetist. It is common for an anaesthesia/surgical team to consist of at least two nurses and one doctor for most invasive and/or critical avian surgeries.

PATIENT RECOVERY

Once the procedure is complete, the surgical nurse should prepare for recovery. Most birds experience some delirium following anaesthesia. This delirium usually occurs shortly after discontinuing anaesthetic gas and can result in vigorous wing flapping. Injectable anaesthetics may produce more severe post-anaesthetic delirium.[6] In preparation for recovery, all monitoring and supportive devices should be removed unless vital to the patient. Also, examine the patient's oral cavity for any regurgitated food and clean as needed. The recovering bird should be lightly wrapped in a towel to control any delirious flapping. When stable enough to stand, place the bird in a warm quiet place until full recovery occurs. Without disturbing the patient, monitor for any regurgitation or other problems that might occur during recovery. Offer food and water as soon as the patient is fully recovered (usually within 30-60 minutes for most routine anaesthetic procedures).[6]

Managing Pain in Avian Patients

Understanding and managing pain in birds is difficult when compared to mammals. Birds clearly experience pain but analgesics are poorly studied in birds, especially psittacine birds.[18,25] It is reasonable to assume that pain can impede avian patient recovery just as has been proven in other animals.[25] Birds may possess different types and quantities of opioid (pain) receptors when compared to mammals.[25] Studies in pigeon forebrains have shown that kappa opioid receptors predominate suggesting that kappa opioid receptor agonists would be appropriate for avian pain control.[8] With the current limited studies, birds seem to require a relatively high dose of selected analgesics to produce 'analgesia,' further supporting that birds probably do have differences in opioid receptors, or respond to pain differently when compared to mammals.[25] For example, umbrella cockatoos had no discernable analgesic response (compared to saline) when given high doses of fentanyl (80-100 times more potent than morphine) and only 4 of 7 had some measurable analgesia when given 10 times the 'high dose'.[26]

The recognition of pain is very important in avian medicine. As is true of many wild animals, birds tend not to show overt signs of pain (vocalizing) as obvious distress behaviors may attract unwanted attention. Even some physiologic measures of pain such as increased heart rate are not always seen in 'painful' birds.[27] As clinicians, we often use our own judgement when evaluating pain in birds. For example, a bird with a recent fracture or just recovering from an invasive procedure is likely experiencing pain. But also watch for birds that are guarding a limb, picking feathers over a limited area, acting aggressive, having focal muscle contractions, shivering, anorexic, or intensively fearful or phobic in response to basic handling or approaches (fear of a person touching a painful area or falling on a sore keel). It is the subtle, and not overt, signs of pain that more likely predominate in painful avian patients.

The first steps to resolving pain are to recognize the source of pain and reduce discomfort. For example, splinting a fractured leg or covering an open wound with a light bandage may instantly reduce pain and anxiety. Other ways to reduce anxiety and pain include placing the avian patient in a quiet, warm environment and removing cage 'furniture' and making food and water easily accessible to decrease painful movements.[27]

Evaluating the response to pain treatment is difficult to assess in birds using the current recommended analgesics. Obvious noxious stimuli may result in a withdrawal response, but this is not true in all cases. In parrots given a mild electrical stimulus to the foot, the expected response was to withdraw the foot and show reluctance to placing it back on the perch (as was noted).[15] However, several parrots in the study began wing flapping, and not foot withdrawal, when the electrical stimulus was given.[15] Chickens display an instinctive behaviour called 'crouching immobility' when exposed to repeated noxious stimulation. This immobility reaction in chickens is prolonged when fear is present and subsequently decreasing the fear component reduces the time of immobility. Pigeons consistently had body trembling for several hours after orthopaedic surgery as compared to controls that received general anaesthesia, opioid analgesia and no surgery. Other signs of pain include weight loss, anorexia, a behavior change, aggression, feather picking over a focal area, guarding to protect a specific site and decrease in social vocalisations.[15]

Despite the uncertainties in bird responses and until additional studies are conducted, pain management should still be encouraged for avian patients. Butorphanol is recommended for severe pain and is given at 1-3 mg/kg IM q 6-24 hrs.[25, 27] Butorphanol given at 1 mg/kg also has isoflourane-sparing effects, in addition to analgesic properties, in studied birds.[27] Butorphanol is a mixed agonist-antagonist with primarily kappa agonist action.[15] Clinically, butorphanol given at 2-4 mg/kg IM or per os seems to be safe and offers relief to some avian patients. Relief may be evidenced as a calming effect on the bird. Again, it is uncertain whether this calming effect is due to sedation from the high dose of butorphanol or true analgesia. Pigeons that receive butorphanol before and after orthopedic surgery return to normal behaviours quicker than those receiving postoperative butorphanol only.[15]

Banamine, given at 1-5 mg/kg IM q 24 hrs, may also provide some analgesia.[18] However, intestinal and renal side-effects from banamine administration have been induced with doses as low as 0.1 mg/kg in quail. Similar renal lesions have been reported in numerous bird species.[27] Aspirin given at 5-10 mg/kg PO q 24 hrs appears to reduce pain. Both carprofen (1 mg/kg SC or 2-4 mg/kg PO q 8-12 hrs) and ketoprofen (2 mg/kg IM or SC q 8-24 hrs) are popular analgesics used in avian medicine.[27] Meloxicam and piroxicam have also be advocated for use as pain control in birds.[15] As the side effects of all analgesics have not been studied in birds, these drugs should be used with care.

Although there are few avian studies, most nonsteroidal anti-inflammatory drugs (NSAIDS) are eliminated by renal clearance and should be used with caution, as they have been associated with a variety of renal lesions in birds and mammals.[28,29] NSAIDS have also been advocated as 'first course of therapy' for managing chronic pain.[15] Flunixin meglumine (Banamine®) induced glomerular lesions in bobwhite quail (*Colinus virginianus*) that increased in severity proportionally with the dose.[28] Aspirin has been associated with significant inhibition of prostaglandin synthesis (specifically prostaglandin F_{2a}) in Japanese quail.[30] In this same experiment, aspirin was shown to induce liver enlargement resulting from hepatic lipid accumulation in N-6 FA deficient birds.[30] Aspirin injected IV into Pekin ducks induced temporary diruresis lasting 30 minutes. This is in contrast to its antidiuretic effect in mammals. Aspirin had no effect on GFR or peripheral blood pressure.[31]

Diclofenac has been identified as the main cause of a 95% decline in the Oriental white-backed (*Gyps bengalensis*), long-billed (*Gyps indicus*) and slender-billed (*Gyps tenuirostris*) vultures endemic to South Asia.[32] Controlled diclofenac toxicity studies have shown that it is highly toxic to all *Gyps* species. Treated birds die within 2 days of a single dose of 0.8 mg/kg PO, with renal failure and visceral gout.[32] As a comparison, the LD_{50} of diclofenac in *G bengalensis* is 98-225 µg/kg whereas the same values for mice and rats are 95,000-1,300,000 µg/kg and 53,000-1,500,000 µg/kg, respectively.[33]

Surveys from veterinarians and zoos documenting use of NSAIDS in over 870 scavenging birds from 79 species have shown similar problems.[34] Birds that showed adverse effects included *Gyps* vultures, birds of prey, owls, cranes, storks and crows. Carprofen and flunixin meglumine were associated with mortality in 13% and 30% mixed avian species, respectively. Mortality, renal disease and gout were also reported with ibuprofen and phenybutazone use. A few instances of toxicity were recorded with ketoprofen use, however, these birds did not die. Of note, meloxicam was used in over 700 birds representing 60 species with no reported mortality.[34] Meloxicam was not found to be toxic in *Gyps* sp at doses that wild vultures would likely encounter.[33]

Local (injected and topical) anaesthetics can be effective for producing regional anaesthesia. The two most commonly used local injectable analgesics, lidocaine and bupivacaine, block nerve axon sodium channels interfering with pain impulses along the nerve.[15] The total dosage of lidocaine should not exceed 2-3 mg/kg. While there is not an established bupivacaine dose in birds, 1 mg/kg total dose has been recommended as safe for 'large birds.' Intra-articular bupivacaine (3 mg in 0.3 ml) was effective in reducing pain in chickens with experimentally induced acute arthritis. Topical bupivacaine applied to the amputation site of beak-trimmed chickens provided 4 hours of analgesia in one study. Duration of action of both drugs has not been established in birds. Noted toxic side-effects include ataxia, fine tremors to seizures, cardiovascular effects, stupor and death.[15]

[a] Veterinary Specialty Products, Inc., Boca Raton, FL

Summary

Avian anaesthesia is significantly different than mammalian anaesthesia techniques. A solid understanding of basic avian anatomy and respiratory physiology is essential for safe anaesthesia of birds. Inhaled gas anaesthetics, such as isoflourane and sevoflourane, are commonly used with birds. Those interested in avian anaesthesia and analgesia are encouraged to stay current as research and new information on this topic is frequently being reported.

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