PRACTICAL AVIAN ANAESTHESIA

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Preanaesthetic Considerations

As for all of our patients, birds should be assessed to determine their likelihood of surviving both the anaesthetic and procedures to be carried out under anaesthesia (see Evaluation of the Avian Surgical Patient by Anne Fowler in these Proceedings). Ideally this would involve complete history, physical exam, CBC and biochemistry. However the stress these assessments place on the patient must be taken into consideration when deciding what a "minimum database" means for each individual case. Basic assessment of body condition, level of hydration, patency of airway, mucous membrane colour and auscultation of heart and lungs should be carried out in all cases. Where necessary the avian patient should be stabilised medically before anaesthesia is contemplated.

Failure to empty the crop prior to anaesthesia can result in regurgitation and aspiration of ingesta. However there is much disagreement as to the length of time birds should be fasted. Due to the rapid metabolic rate and poor hepatic glycogen storage of birds, fasting for more than 2-3 hrs is generally not recommended. However it does not appear to be harmful to remove food overnight and water 2-3 hrs prior to anaesthesia in birds larger than cockatiel size as long as they are in good body condition.[1] In all cases the crop should be palpated and emptied of contents prior to induction of anaesthesia.

Dehydration can be a significant problem in the avian patient. Balanced electrolyte fluids should be considered even for healthy birds undergoing non-invasive procedures. Where there is a potential for blood loss leading to hypovolaemia subcutaneous (SC), intravenous (IV) or intraosseous (IO) fluids should be administered. Warming these to approximately 39^{o}C may also help maintain body temperature. Lactated Ringer's Solution (LRS) is generally recommended and can be given at 5-10ml/kg/hr IV or IO. Shock doses can be given at 30ml/kg/hr in most birds. [2]

Birds do not show pain in overt ways as most predatory mammals do. This does not mean that they do not perceive pain and in any potentially painful procedure analgesia should be administered pre-emptively. Pharmacodynamic studies in pigeons suggest that birds respond better to butorphanol than morphine, buprenorphine or fentanyl.[3] Butorphanol also lowers the heart rate, tidal volume and expiratory/ inspiratory times and reduces minimum anaesthetic concentration in some parrots.[3] Administration prior to induction of anaesthesia and/or post operatively is preferred as anecdotal reports suggest intraoperative administration can result in a sudden increase in depth of anaesthesia making monitoring more difficult. Butorphanol is commonly used at 1-3mg/kg IM q4-6h.[4]

Hypothermia is associated with decreased cardiac stability, increased anaesthetic sensitivity and prolonged recovery time. Birds are more susceptible to hypothermia during anaesthesia because of: their relatively small body mass in relation to surface area, removal of insulating feathers for surgical preparation, application of disinfecting fluids, the drying effects of anaesthetic gases and loss of the physiologic ability to maintain body temperature while anaesthetised. Heat loss can be reduced by minimising the amount of feathers removed, operating in a warm room, using heating devices such as heating pads, warm towels, circulating water blankets, warm air blankets or using warmed IV fluids. It has also been shown that warming and humidifying the medical oxygen used to deliver anaesthetic gases significantly reduces the loss in body temperature.[1]

Induction

Mask induction, using a non-rebreathing system, is generally the most practical method used for companion birds. The bird is gently restrained to avoid flapping and injury to the wings while the head is placed completely within the mask. Care must be taken not to damage the beak or eyes. Isoflurane is generally used because it is relatively low cost, has comparatively rapid induction and recovery, low blood solubility and minimal metabolism.[1] To achieve rapid induction isoflurane is administered at 4-5%. As soon as anaesthesia is achieved vaporiser settings can be reduced to minimum anaesthetic concentration for maintenance.

The advantage of non-rebreathing systems include a rapid response to changes in vaporiser setting and a lower resistance to breathing.[1] Oxygen flow rate in these systems should be approximately 2-3 times the minute ventilation or 150 to 200ml/kg/minute.[5]

The size and shape of mask used is dependent on the size and shape of bird. Normal small animal anaesthetic masks can be used. An inexpensive alternative is to use cut down bottles or syringe covers with a latex glove or cling wrap covering one end. A hole approximately the diameter of the bird's neck should be made in the latex glove to allow a good seal around the neck. This minimises loss of anaesthetic gases which can result in slower inductions. Some species, especially waterfowl have a "dive response" which results in breath holding when the beak is held closed. In these species, and birds where blockage of the nostrils may be a problem, care should be taken to choose a mask large enough to allow open mouth breathing.

Injectable Agents

The disadvantages of injectable anaesthetic agents include significant species variation, cardiopulmonary depression, prolonged and violent recoveries. These disadvantages generally mean that injectable anaesthesia is not favoured over inhalant anaesthesia where there is an option. [1, 10]

Diazepam and midazolam can be used to sedate fractious patients prior to mask induction. Neither has any analgesic effect. Uptake of Diazepam, when given intramuscularly, is slow and unpredicatable. Midazolam is more potent and longer lasting than Diazepam, with rapid intramuscular uptake. In some species e.g geese, raptors, pigeons, its effects may last several hours after anaesthesia.[1]

Xylazine and medetomidine can be used alone or in combination with ketamine. They can have profound cardiopulmonary effects in some species. When used alone, xylazine at high doses is associated with respiratory depression, excitement and convulsions. Overdoses or slow recovery can be treated with reversal agents such as yohimbine or atipamezole. [1]

Ketamine has been used where restraint and moderate analgesia are required for minor surgical and diagnostic procedures. It may be used alone or combined with diazepam or xylazine to improve muscle relaxation or increase analgesia. There is however significant species variation in its effectiveness.[1] Waterfowl generally do not do well with ketamine/xylazine combinations.[4]

Propofol can be used for induction and maintenance however it needs to be administered intravenously and can have prolonged recovery times and rough recovery compared to isoflurane.[1,10] Short periods of apnea and cardiac arrhythmias following induction have been reported. [4,12]

Maintenance

When performing invasive procedures and where the procedure is likely to last more than ten minutes birds should be intubated if possible. In most birds greater than 100g this is not difficult as the glottis is readily visible at the base of the tongue. In parrots and other fleshy tongued species the tongue may need to be gently grasped and pulled forward to visualise the glottis.

As birds have rigid, non-expandable tracheal rings small un-cuffed endotracheal tubes should be used to reduce the risk of damage to the tracheal mucosa. Tube size should be chosen to achieve a good seal with the glottis without a tight fit. However it is also important to note that in some species the tracheal lumen diameter deceases caudally. These birds require a tapered endotracheal tube or to use a narrower tube than the glottis diameter would suggest in order to reduce the chances of tracheal mucosal damage resulting in stricture.

In birds less than 100g, catheters can be used as endotracheal tubes. Small diameter endotracheal tubes have an increased resistance to air flow and therefore increase the effort required for spontaneous respiration. Also the drying affects of anaesthetic gases increase the tenacity of respiratory mucus resulting in a greater risk of narrow endotracheal tubes becoming blocked by mucus plugs. Hence it is sometimes better to maintain small birds on a mask.

Some birds, such as penguins, pelicans, kiwi and some ducks have a ventral projection in the lumen of the glottis that makes intubation more difficult. These birds will require a smaller endotracheal tube or to be maintained with a mask.

It is also important to remember the “dive response” mentioned earlier when securing the endotracheal tube. These species will not breath spontaneously if the beak is taped shut – tape the tube to the lower beak only or around the back of the head.

There is much species variation in the length and shape of the avian trachea. In species with long curving trachea the endotracheal tube should sit no further than just caudal to the glottis to reduce the risk of blockage due to kinking of the trachea against the end of the tube. In the “typical’ bird , and certainly in most companion species, the trachea is longer and wider in comparison to body size than in most mammals. This results in increased tracheal dead space. Birds compensate for this by having a slower, deeper respiratory pattern.[1]

In mammals inspiration is an active process requiring the contraction of inspiratory muscles. Expiration however is a passive process requiring no active muscle contraction to decrease intrathoracic volume.[6] In birds, both inspiration and expiration require muscular activity. Contraction of the inspiratory muscles moves the ribs cranioventrally resulting in increased internal volume of the coelomic cavity and negative pressure in the air sacs relative to ambient atmospheric pressure. This leads to air flow through the rigid lungs (across the oxygen exchange surfaces) and into the air sacs where no oxygen exchange occurs. With contraction of the expiratory muscles, moving the ribs caudodorsally, air flows from the air sacs back through the lungs (across the oxygen exchange surfaces again) and out the trachea.[1,11]

When the bird is positioned in dorsal recumbency abdominal viscera compress the abdominal air sacs resulting in decreased volume of gas and reduced ventilation. Hypoventilation results in hypercapnia within 15-20 minutes and eventually leads to apnea.[4] Respiratory gas volume per unit body mass of the avian respiratory system is 2 - 4 times that of the dog. However only 10% of the total specific volume is contained in the “oxygen exchange” areas of the avian respiratory system compared with 96% in the mammalian lung. This means that apneic periods are more critical as there is significantly less residual functional capacity.[1]

Intermittent Positive Pressure Ventilation

Intubation and the provision of intermittent positive pressure ventilation will help maintain a normal physiologic state. This can be achieved by either manual compression of a rebreathing bag or use of mechanical ventilators. When performing manual positive pressure ventilation the rebreathing bag should be compressed every 30 seconds with sufficient pressure to visibly expand the chest as much as would be expected during normal conscious respiration.

Mechanical ventilators come in two types – volume-limited and pressure-limited. Volume-limited machines deliver a preset tidal volume regardless of airway pressure. Pressure-limited machines deliver a tidal volume until a preset airway pressure is reached. Studies in mammalian models have shown that pressure-limited systems compensate for air leakage better than volume –limited systems.[7] This would suggest that pressure limited systems are best for our avian patients as during intra-coelomic procedures we often open one of the caudal air sacs allowing gas leakage. However the pressure limited system also has its limitations. If the airway were to become occluded the machine will deliver a lower tidal volume for the same airway pressure.[1]

The author is familiar with the Vetronics SAV03, a pressure cycled ventilator designed for use on intubated animals up to 10kg in weight. It is designed to be used as a T piece, non-rebreathing system with a simple switch changing operation from spontaneous breathing to positive pressure ventilation. Using the incoming gas from the anaesthetic machine, this system controls the gas delivered to the patient in the inspiratory phase until a set airway pressure is reached. At this point the patient is allowed to exhale. A user-defined delay then elapses before inspiration begins again and the cycle repeats. The peak airway pressure can be adjusted to control the depth of respiration. Valve status indicators show whether the patient is in an inspiratory or expiratory phase. These can be monitored to watch for shortened inspiratory time which may suggest occlusion of endotracheal tubes or loss of respiratory compliance leading to hypoventilation. When there is leakage from an opened air sac it will take longer to reach peak airway pressure (longer inspiratory phase) resulting in continuous flow of gases over the oxygen exchange surfaces and easier maintenance of anaesthesia (more information about this system can be found at http://www.vetronic.co.uk). To avoid trauma to air sacs, airway pressures should not exceed 15-20cm H_2O when using IPPV.

Air Sac Cannulation

Where the trachea is occluded or surgical access to the oral cavity or head is required, an air sac cannula can be placed in the caudal thoracic air sac to maintain anaesthesia.[1, 2, 4, 8] Where possible anaesthesia is first induced using a mask. The patient is then placed in right lateral recumbency with the dorsal leg pulled either cranially or caudally. [2,9,13, 14] The landmarks for placement of the incision have been listed as between the 7th and 8th rib [9], in the paralumbar fossa caudal to the 8th rib [2, 13] and where the last rib passes under the flexor crura medialis muscle.[14] A small skin incision is made and right angle forceps or scissors are used to push through the abdominal wall muscles into the underlying air sac. A shortened endotracheal tube, or similar, of approximately the same diameter as the patients tracheal lumen is then placed into the air sac. A small feather can be held over the tube opening to check for air movement indicating correct placement into the air sac. Once placed the tube is sutured to the skin to hold it in place.[2, 9,13]

Placement of an air sac tube introduces bacteria and rapidly induces inflammation. Hence antibiotic and antiinflammatory cover should be considered when air sac cannulae are used. Air sac cannulae should be removed within five days of placement.[9]

Monitoring Anaesthetic Depth

Wherever possible anaesthesia should be monitored by a well trained nurse or technician so that the surgeon is free to concentrate on surgery. This allows for faster more efficient procedures therefore reducing the risk of side effects due to prolonged anaesthesia.

Many of the reflexes used for monitoring anaesthetic depth in mammals can also be used in birds but may not be as reliable in some avian patients. When a surgical plane of anaesthesia is reached the patient's eyelids are completely closed, the pupil is mydriatic, the pupillary light reflex is delayed, the nictitating membrane moves slowly over the entire cornea, the muscles are relaxed and all pain reflexes are absent.[1] As the patient becomes deeper under anaesthesia these reflexes are lost and breathing becomes slow and shallow. Sudden piloerection and pupillary dilation usually occur with cardiac arrest.[15]

The most common problems during anaesthesia are apnoea, hypoventilation, hypothermia and regurgitation.
The only way to accurately assess ventilatory status of a patient is by measuring arterial CO_2. However this is not practical in most general practice situations. One study has shown capnography can be used effectively to estimate this parameter. This also has its limitations however and methods that allow reduction of endotracheal tube dead space must be used.[1]

Where capnography is not available observations of respiratory rate and depth remains the best manual assessment method. The patient's respiratory pattern can be monitored by watching the patient directly, if using transparent surgical drapes, or watching the movement of the rebreathing bag. In very small patients use a party balloon for a rebreathing bag to make movement more visible. If using an IPPV unit such as the Vetronics system mentioned earlier the respiratory phase indicators can be watched to monitor rate of respiration.

Pulse oximeters have not been shown to be consistently accurate in avian patients.[1]

Heart rate and pulse strength can be monitored using direct auscultation or palpation where the patient is large enough to allow access by the nurse anaesthetist without interfering with the surgeon. ECG can be used, however some ECG units are not capable of detecting and accurately recording the rapid heart rates of some avian patients. To avoid damage by alligator clips attach ECG leads to 27 - 26g needles or sterile wire sutures and place the needle/ suture through a thin piece of skin on the wing or leg. ECG readings in birds are interpreted differently to mammals due to differences in the direction of the RS segment.[2]

Recent work with Doppler flow ultrasound has shown that Doppler units can be reliably used to monitor heart rate and blood flow.[1] When placed over a peripheral artery such as one of the visible vessels crossing the medial elbow they can be used to give an audible sound with each heart beat. They have also been used in conjunction with small blood pressure cuffs placed over the distal femur or humerus to collect blood pressure information. When systolic pressure drops below 90mmHg bolus doses of crystalloids at 10ml/kg and colloids at 5ml/kg can be given IV or IO until systolic pressure returns to above 90mmHg.[16]

Temperature can be reliably monitored using oesophageal probes placed to the level of the heart.[1] This requires careful manoeuvring of the probe through an empty crop to the thoracic inlet. Cloacal temperature monitoring is not as accurate and dependent on body position and cloacal activity.

Recovery

Once anaesthetic gases are turned off recovery from anaesthesia is generally rapid. The anaesthetic circuit should be disconnected and flushed then reconnected and the bird allowed to spontaneously breathe 100% oxygen while recovering. When the bird starts to show signs of jaw movement it should be extubated. Gently wrapping in a soft cloth or paper towel will help prevent flapping or sudden movement that may cause injury. The bird should be held or placed in an upright position until it is able to hold its head up and then placed in a warm dark cage, box or humidicrib. Most birds will be fully recovered within 20minutes.

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