Basic Avian Surgery

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A basic understanding of general surgical principles should be understood prior to avian surgery. Although there are many anatomical and physiological differences between birds and mammals, surgical techniques are very similar. Due to small patient size and anatomical differences (avian air sacs for example), microsurgical instrumentation with magnification with focused light are often necessary for efficient bird surgery. Because of physiologic variations (compared to mammals, birds exchange oxygen on inspiration and expiration and can frequently go into cardiac arrest following relatively brief apnea), anaesthetic techniques in avian species are very different and are discussed at another session during this conference.

Doolen listed several principles that hold very true to maximize avian surgical success.[1] The first is to minimize hemorrhage. The second is to minimize tissue trauma. Third is to minimize anaesthetic time. Fourth is to minimize anaesthetic and metabolic complications. Last, provide post-surgical support and analgesia. These seem simple enough, but are very important to understand and practice during all avian surgical procedures.

If interested in avian surgery, actively pursue continuing education. One of the best continuing education courses is at one's own hospital in the form of necropsy patients. If permitted by the owners, perform as many necropsies as possible to gain experience and exposure to avian anatomy, tissue handling and microsurgical instrument use. Also attend continuing education courses conducted by the Association of Avian Veterinarians, American Association of Zoo Veterinarians or other groups supporting avian medicine and surgery. Several texts listed in the 'Recommended Reading' section at the end of this paper give excellent descriptions of numerous avian surgeries. Publications such as the *Journal of Avian Medicine and Surgery* provide numerous well-referenced papers on surgical techniques, in addition to medical subjects.

Also familiarize yourself with the numerous potential surgical 'tools'. These 'tools' include radiosurgery, microsurgical instruments, endoscopes, high powered microsurgical loops with light, operating microscopes, laser units and numerous other items that have become commonplace with avian surgery. Consult with surgical instrument companies, colleagues and the continuing education resources listed above.

During the presentation, the author will discuss his most commonly used instruments and 'tool's used with avian surgeries. Although little information exists on suture material in birds, chromic catgut, polyglactin 910, polydioxanone (PDS), monofilament nylon and monofilament stainless steel have been evaluated in rock doves (*Columba livia*).[2] From this information and the author's experience, PDS is slowly absorbed and causes minimal tissue

reaction making it suitable for both internal and skin closure use.[2] For the purposes of this discussion, PDS will be used for all monofilament, absorbable sutures in bird surgeries.

Abdominocentesis

Abdominocentesis is indicated for birds with free abdominal (peritoneal) fluid of unknown type or origin. Due to the coelomic, intestinal-peritoneal and dorsal and ventral hepatic-peritoneal cavities in normal birds, a single free 'cavity' does not exist in the avian abdomen. Attempts at 'abdominal washes' are difficult and can result in fluid infusion into the abdominal or caudal thoracic air sacs. With increasing abdominal fluid, the avian air sacs become compressed, the abdominal wall distends and the patient may develop varying degrees of dyspnea. An affected patient should be restrained with the long axis of its body oriented vertically, or slightly tilted forward, and head up. Due to potential respiratory compromise, the use of anaesthesia may be contraindicated.

The ventral abdomen is aseptically prepared and a small gauge needle (21 g to 27 g) is inserted on the midline just caudal to the sternum (keel). In effort to avoid the ventriculus (gizzard), the needle is directed to the right side of the body and any fluid present is then aspirated. As with other avian fluids and if needed, process the aspirated sample(s) quickly and use for cytological, microbiological and biochemical analysis.

Bone Marrow Aspiration and Intraosseous Catheterisation

Bone marrow biopsy in birds may be indicated in cases of unexplained anemia, heteropaenia, leukopaenia, pancytopenia, thrombocytopenia, abnormal peripheral blood cells and suspected hematopoetic neoplasias and bone marrow infections. The proximal tibiotarsus and the sternum (keel) are the most commonly discussed sites for collecting avian bone marrow aspirates and biopsies. All biopsy sites should be aseptically prepared prior to collection and ideally the patient should be anaesthetised. Pediatric Jamshidi bone marrow biopsy, Jamshidi Illinois-Sternal/Iliac aspiration, spinal and standard needles may all potentially be used. Size of the biopsy site, sample needed (aspiration cytology v. biopsy) and clinician experience will dictate the needle type and gauge required.

The proximal tibiotarsus is frequently used for bone marrow collection in birds. The following description also applies to intraosseous catherization. With the patient in dorsal recumbency, the chosen leg is extended and flexed at the stifle. With one hand, stabilize the bird's leg and determine the orientation of the tibiotarsus and its location within the surrounding leg muscles. Using the free hand, aseptically insert the needle into the stifle just lateral or medial to the patellar tendon. While aligned with the diaphysis, gently rotate and advance (from proximal to distal) the needle through the flattened cranial portion of the proximal tibiotarsus into the marrow. Advance the needle approximately one third to one half the length of the tibiotarsus. Confirm entry into the bone marrow by rotating the needle, pivoting at the proximal tibiotarsus and causing the end (of the needle) to 'scrape' the medial side of the cortex and associated bony spicules. This confirmation technique is both audible and palpable and seems to increase cellular yield (possibly by dislodging bony spicules), especially when only aspiration cytology is used. When placing an intraosseous catheter, this 'scraping' technique is not necessary

An alternate location for intraosseous catheterization is the ulna. Aseptically prepare the dorsal carpus. Flex the carpus such that the flattened distal end of the ulna is palpated. Advance a needle through the skin into the distal ulna in the direction of the diaphysis. The needle should be advanced through ½ to 2/3 the length of the ulna. Confirm placement by administering isotonic fluids. If correctly placed, the basillic (wing) vein, which crosses the ventral elbow, will quickly fill with fluid as it is being administered.

Although the sternum marrow compartment may be too small in some species, as with psittacines and raptors, the keel is another acceptable site for bone marrow collection. Additionally, this site may be needed when the tibiotarsus is unavailable for some reason. With the patient in dorsal recumbency, the widest portion of the keel is chosen and a small stab incision is made through the skin. Aspeptically advance the needle through and parallel to the center of the keel, which is perpendicular to the flattened portion of the sternum, and into the marrow.

Misdirected needles may penetrate through the cortex and into surrounding soft tissues. Simply redirect the needle if no marrow was obtained. Also, some birds undergoing polyostostic hyperostosis (osteomyelosclerosis) are difficult to obtain bone marrow samples from. During polyostotic hyperostosis, up to three fourths of the medullary cavity's hematopoetic tissue can be replaced by medullary bone.[3] In cases where bone marrow samples are difficult to obtain (ie. increased medullary bone, medullary sclerosis, etc), biopsy, over aspiration cytology, or choosing alternate sites would be indicated.

Once in either the keel or tibiotarsus marrow, use a syringe to provide negative pressure until marrow appears in the hub of the needle or tip of the syringe. Avoid excessive or prolonged negative pressure to prevent peripheral blood contamination. To prevent plugging with instruments that have no stylet (plain needles), use a small Steinmann's pin to create an opening into the marrow cavity before placing the needle. Alternatively, the plugged needle is pulled and replaced with a new instrument. Once an adequate sample is collected, remove the needle and process the marrow immediately.

Use an air filled syringe to expel the marrow immediately, from the needle, onto a glass slide. If cultures are needed, the collected sample should be transferred directly to a sterile swab, culture plate or media and further handled. Due to the fragility of avian blood cells, the 'two cover glass' technique has been recommended. Marrow often appears as pale, fatty or gritty and can be used as quick visual aid to help confirm proper collection.

Crop Biopsy and Repair

Crop biopsy may be indicated for determination of crop masses (most commonly papillomas), screening for proventricular dilatation disease (PDD) and any other suspected abnormalities where histopathologic evaluation is required. In two separate studies, crop biopsy was reported to be 68% and 76% sensitive in detecting PDD in psittacine birds.[4, 5] In the author's experience, crop biopsy is a relatively poor screening tool for PDD. Regardless, a normal crop biopsy does not rule out PDD. Incise the skin over the left side of the crop near the thoracic inlet. Bluntly separate the skin and crop until you can pull the crop partly out of the incision. If screening for PDD, remove a large 1-2 cm section of crop including large blood vessels.

For other samples, remove the abnormal tissue or masses present. A two layer closure works best with crop incisions. The first layer is closed with an inverting suture and the author closes the skin and crop together as the second layer. Monofilament absorbable suture is ideal. The same approach is used to retrieve crop foreign bodies and to perform proventricular and ventricular endoscopy.

Crop repair is most often indicated following thermal burns in young. Following the thermal burn and prior to surgery, wait until the margins of the necrotic tissue are clearly visible (usually 4-7 days after the incident). Remove all necrotic tissue and close as described above. The crop has an incredible ability to stretch and even large crop resections seem to be well tolerated by most young birds. Subsequent feedings will obviously need to be reduced depending on the post-operative size of the crop.

Pharyngostomy Feeding Tube

Pharyngostomy feeding tubes are used in birds to bypass the upper gastrointestinal tract (as with oral or upper cervical esophageal disease) and/or to provide alimentation to the crop or proventriculus of anorexic patients. With the patient anaesthetised, place a moistened cotton tip applicator into the cervical esophagus per os. Use the applicator to 'push' the esophagus to the right and laterally. At approximately the mid-neck region, make a 1-3 mm stab incision over the applicator with just enough pressure to incise the skin and right lateral wall of the esophagus. Exteriorize the opened esophageal wall and advance a red rubber (or similar) feeding tube into the lower esophagus/crop region. If needing to bypass the crop, the feeding tube can be gently advanced further into the proventriculus. Although dependent on patient size, most birds can safely take a 12-16 French red rubber feeding tube. Once in place, suture the tube to the esophageal mucosa and then to the overlying skin. Pharyngostomy feeding tubes can remain in place for several days or longer if needed. Pull the tube when no longer needed and allow the wound to heal by second intention.[6]

Skin Biopsy

As most avian skin contains a normal, surface, bacterial population, obtaining meaningful cultures requires some special techniques. Microscopic cytology or histopathology may help support the diagnostic value of skin cultures. Direct skin surface cultures often result in environmental and 'normal' skin microorganism contaminates, necessitating more invasive methods of diagnostic collection.

Preferably, surgically exposed dermal or subcutaneous tissue is sampled for microorganism evaluation. If biopsying the skin, aseptically sample both feather pulp and subcutaneous tissue in multiple sites. Of the two, subcutaneous cultures are more meaningful while feather pulp tends have low diagnostic yield. With a small section of skin removed for biopsy, gently lift one of the skin margins and use a small swab (ie: Mini-Tip Culturette) to sample deep under the epidermis without touching the surgical instruments or overlying skin. If present, multiple feathers, especially pins, should also be cultured and included in the skin biopsy. A feather is plucked from the affected skin and cleaned at its proximal end with alcohol. The externally 'sterilized' feather is split at the intradermal calumus (proximal end of feather) and the dermal pulp (inside of the feather) is aseptically cultured. If dermatophytosis is suspected and in

addition to cultures, scraped skin and crushed feather follicles can be treated with sodium hydroxide and then Gram's or acid-fast stained to help identify causative fungal organisms. Quill mites may be diagnosed by splitting the calamus (shaft), placing the contents on a glass slide and soaking in a drop of chlorlactophenol, slightly heating the sample and then viewing the parasites under magnification. The rachis, barb and barbules may also be cultured, without 'sterilization', as has been done to identify saprophytic fungal growth on grossly discolored feathers. Feather pulp cultures tend to have a low diagnostic yield.

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Recommended Reading

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