Practice Tips in Reptiles

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Handling and restraint of reptiles and amphibians

One should NEVER assume that because someone owns an animal – that they know how to properly handle it. Therefore, ONLY You and your hospital staff should be involved in the proper restraint of the animal being examined.

With most exotic species, I find that with experience and proper preparation will allow for the proper handling of most reptile and amphibian patients; this is, for the most part, is usually not very difficult – though one should always be prepared for the unexpected.

Amphibians

In general, with most amphibians, to avoid stress to the animal and any potential injury, handling should be kept to a minimum. I first evaluate the animal in its' enclosure – noting appearance, position, posture, color(s) etc… If I am unable to evaluate the animal well in the owner's enclosure – this is not possible, the patient can be transferred the patient to one of our clear plastic containers (either with or without water depending on the species). This allows me to evaluate of externally many aspects of the animal (external, mostly) without actually handling the patient. I always use good lighting and magnifying devices to scan the entire amphibian, including trans-illumination the animal though the clear plastic. Once I have evaluated the animal the patient has been evaluated without restraint, (again all the while discussing the visual exam & what I am doing with the client and asking more questions), the patient is ready to be handled. – I then am ready to handle the patient.

Amphibians produce various secretions (i.e. mucous) to protect themselves against opportunistic pathogens. Disruption of the skin secretions from abrasions caused by mishandling or due to the skin drying out may allow other infectious agents to gain entry into the skin. Therefore when handling amphibians I always use rubber / latex gloves, and rinse the gloves to remove the powder or any other coating on the rubber. If you it is have difficult toy catching the animal(s), fine mesh nets may be used to capture the amphibians. Be careful! - wet amphibians in rinsed latex gloves are slippery. Large toads, giant salamanders and hellbenders should have their heads properly yet gently restrained as they may bite and though it is not often very painful it can be embarrassing. I again completely survey the exterior of the animal using again use magnification as well as trans-illumination as needed to continue my exam. I then palpate the entire animal assessing all aspects from one end to the other. To evaluate the oral cavity, cavity I can at times stimulate the animal to open its mouth by gently pressing on the jaws at the soft tissue of the commissure. If this technique is ineffective, I may have to use a soft clean rubber spatula may be used to gently open the mouth to examine the oral pharynx and glottis. At this point, samples can be collected from the posterior naris and nasal passage. Using a mini-tip culturette samples for cytology and culture can also be collected from the trachea by passing the small swab through the glottis. After I have completed my the physical examination in the room with the client watching, and I have asked all my questions concerning the animal – I indicate that I am going to take the animal back to collect the necessary laboratory samples as well as to measure the body weight and compare it to any previously recorded weights.

Iguanas and other lizards (I usually use an appropriately sized towel for each animal)

In general most lizards are easily restrained – though one does have to be careful with larger species and the potential for injury as well as with smaller specimens and the potential for easy escape. Small lizards may demonstrate the 'fight or flight' response and if not properly restrained may be able to get free - possibly injuring themselves in the process, losing their tail in those species that experience tail autotomy, or worse case scenario – escape somewhere into the clinic. Several defense mechanisms can be seen in all sized lizards but prove to me most potentially hazardous in larger species. Tail whipping is quite common in several species (particularly iguanid species and varanid lizards). If a single holder technique is used, you, the doctor are doing the restraint (single holder) then I typically it is a good idea to keep the tail tucked under my arm, or immobilized between the holder myself and the table, (or even at times between the holder's my legs). I always use a towel that is long enough to surround not only the animals' body but the tail as well. This helps to keep the animal controlled/immobilized. One can then unwrap the areas for evaluation as needed. With two holders, – again I wrap the entire animal and the tail completely in a towel and then while the animal is properly restrained by the handler I can then unwrap the section to be examined. This allows for the patient to be under control during the entire exam, all the while taking practicing great care to avoid any/all possible injury to any part of the animal (and especially the tail).

Biting is another response that I have seen both used defensively as well as aggressively. Proper control of the head and more specifically the mouth is critical to avoid any and all possible injuries. The head may be controlled by placing your hand(s) behind the mandible and around the neck. The holder must be careful not to damage the large dorsal spines in those species where they are present, i.e. adult male iguana's. Using the towel mentioned also helps provide a layer of protection between the handler you and the lizard – especially in those very large specimens.

I recommend never reaching over and or in front of the mouth of a lizard that is not being properly restrained – for though most often they only feign a bite – there are times when animals that can (and will) inflict serious injury. This should also be carefully explained to your clients – that they should never be allowed to in any way get themselves close enough to their pet while it is being examined that they might end up being injured. To evaluate the oral cavity of lizards I can at times stimulate the animal to open its mouth by gently pressing on the jaws at the soft tissue of the commissure or by. I can also gently pulling down on the tissue below the mandible (which in some species is anatomically called the dewlap). I find that slow gentle pressure on the dewlap often results in the animal slowly opening its' mouth. In lizards and crocodilians I have also used the vagal-vagal response may be used to help facilitate the animals to "relax". The vagal – vagal response is when pressure, often digital, is applied to both eyes for a period of time, up to a few minutes. It has been shown that some patients will respond with a decrease in heart rate and blood pressure – and this will often result in them becoming more sedate16sedate2. Though I have found that Even after this vagal sedation has been accomplished, one has to be vigilant for the patiently may be aroused from this state by physical stimulation or noise. If these techniques do not work – or do not work completely - then I may have to use a soft clean rubber spatula may be used to gently open the mouth to examine the oral pharynx and glottis.

Though lizards with long or sharp nails can scratch – typically the nails are not used as a defensive or aggressive mechanism. None the less, in an ideal world, the owner/client would have clipped all the nails prior to exam. To avoid any possible scratches to those working with the animal it is common practice in the author's practice to commonly provide a complementary nail trim on those lizards being examined. In handling old world chameleons – (these lizards typically are most at rest when all four legs as well as the tail are securely attached to something – preferably a branch. So during the physical exam – incorporation of a suitably sized perch to allow the animal to rest during examination may be beneficial. When directly handling these lizards, – I typically allow them to hold onto the handler's my hand with all their feet &and tail and examine and manipulate one leg at a time while the others remain attached to the handler. In those very large lizards that are not being very cooperative with regards to your physical examination, the best your defense is to control head, body and tail. Again, the head can be restrained by holding firmly behind the mandible and around the neck with the thumb and first finger(s). To control front feet and arms hold them gently yet securely against the body of the lizard. To secure the rear legs if needed they can be positioned and grasp up against the tail just below the pelvis. And the tail should be tucked under the arm or between the holder and the table or wrapped in the appropriate sized towel.

Crocodilians

As above for lizards, however, often more handlers may be required depending on the size & and strength of specimens, and the procedures to be preformed. The head and mouth as well as the tail should always be controlled. The muscles used to open the jaws are by comparison relatively weak (to the strength of the biting or jaw-closing muscles) so the mouth can often be easily held closed. It is best for all involved if the mouth of every examined crocodilian and for the safety of all involved is gently though properly taped closed.

Snakes

I always approach any / all snakes slowly and with respect. Though I will say that most are very amenable to handling and so long as the head and body are properly supported – snakes typically do not resist. I find that if snakes that are not correctly supported and or restrained too tightly that they will then struggle. Snakes commonly will try to wrap the end of their tail around you or around the table or other furniture. It is best not to let this happen, as it is difficult to get them to release due to the fact that they often tighten up as you pull on them and they possibly can be injured in the untangling process. Some snakes if ill, threatened, or are in a pre-shedding or/shedding cycle, etc. will may strike out. It is important to locate the head and gently immobilize the head holding gently but firmly just behind the mandible.

To evaluate the oral cavity of snakes, as with lizards, I can at times stimulate the animal to open its mouth by gently pressing on the jaws at the soft tissue of the commissure. I can also gently pulling down on the tissue below the mandible. Here again, I find that slow gentle pressure on this skin often results in the animal slowly opening its' mouth. When examining a snake it is usually a good idea to not try and completely prevent movement by the animal, rather to allow movement of the body with only slight resistance while maintaining control of the head when examining a snake patient. If a snake is not threatening at all, – the head may not need not be completely restrained to evaluate the entire animal. A snake has only one occipital condyle, so dislocation at this joint will occur more easily in snakes than in other species if the head is held too firmly – especially if the snake is fighting to get free.

Concerns about constriction are over-rated as a defense mechanism propagated by bad Hollywood movies. Snakes will constrict if they are threatened, if they think something is a prey item, or if they feel like they are not supported well. However when dealing with very large snakes one needs to have the required amount of help to properly and securely restrain the animal. Bottom line to properly examine a snake it needs to be supported the snake well. Hold the snake's head should be held gently but firmly as required depending on the temperament of the animal. To properly support the snake along its entire body may require using multiple handlers if necessary. Since the vertebrae extend along the entire body, the weight of the snake hanging could potentially dislocate vertebrae or could cause the snake to fight for balance/support. Hold the head gently but firmly behind the mandible and move your hand with the movement of the snake while maintaining control of the head.

Turtles and tortoises

Most Tortoises are docile and relatively easy to handle and examine. They do have the ability to bite quite firmly and can be very reluctant to let go once they bite. They have no teeth but the beak is made of keratin and can be quite strong and sharp. Aquatic chelonians may be more likely to bite &and scratch so additional care should be exercised with these animals. In certain species – especially the male aquatic turtles – the nails can be quite long and therefore can result in scratching. If possible and with the permission of the owner, these nails can be clipped before too much handling to avoid being scratched.

All turtles &and tortoises possess strong legs that can be powerful enough to pin / pinch your fingers between the shell and their legs, and along with being quite painful this situation can also be quite embarrassing when you cannot free your digits. Care must be taken as fingers can be easily trapped against or pulled up against the spurs and sharp pointed edges of some shells as one tries to pull out legs for examination, venipuncture and injections. When attempting to evaluate the oral cavity – one need to first extend and restrain the head. Typically I or an assistant, depending on the size of the turtle, will try to gently immobilize the exposed / extended forelimbs – either holding them directly of pinning them against the shell of the animal. The head is then gently exposed and held just behind the mandible at about the level of the ears using the thumb and forefinger(s). In the case of more resistant turtles I can insert a pair of closed hemostats under the horny upper beak in the area of the premaxilla and carefully lever the head out into a position where I can restrain it. One has to be very gentle when applying pressure on this premaxilla area as the beak – especially in unhealthy turtles can break. Though it will most likely grow back completely – it will be a temporary abnormality that may bother the turtle and perhaps the owner even more. No question in certain turtles and especially the larger tortoises – some form of injectable chemical restraint may be required. Trying to mask / box down a turtle to allow for a complete physical examination may take hours and is typically not advised. To evaluate the oral cavity of turtles, as with snakes, I can at times stimulate the animal to open its mouth by gently pressing on the jaws at the soft tissue of the commissure. At times, in large enough animals, I can also gently pull down on the tissue below the mandible which and I find that slow gentle pressure on this skin often results in the animal slowly opening its' mouth. Like lizards and amphibians, a disturbed, frightened turtle/tortoise may often urinate and sometimes defecate as you pick them up/examine them. One must always be ready for this to happen – 1. to avoid being the target and 2. to be ready to collect possible diagnostic samples. Your defense in examining turtles / tortoises is to hold them midway between the front and rear legs. Keep the animal held out away from your body and watch out for urination/defecation. If possible, clip nails prior to procedures/examination. As in lizards, do not reach across or in front of an unrestrained turtle/tortoise. And as mentioned with all reptiles/amphibians, sedation or anesthesia may be needed, especially in larger, stronger animals.

Review of clinical &and diagnostic techniques in reptiles & amphibians

Sites for and methods of venipuncture

The indications for blood collection in reptiles and amphibians & amphibians are the same as for domestic animals. Sedation may be necessary for intractable specimens. Lithium heparin is the anticoagulant of choice. One should pre-heparinize the syringes prior to venipuncture to avoid blood coagluation in the syringe - due to the fact that it may take some time to collect an adequate volume of blood. One should determine beforehand with the specific diagnostic laboratory being used exactly how much blood they need for specific testing. Generally, for most labs, a volume of o.5 ml of whole unclotted blood is adequate to perform hematology and plasma biochemistries. It is believed that the maximum amount of blood that can safely be taken from a reptile or amphibian is calculated by using the formula: weight (grams) x 1% = ml of blood. For example, an 8.o gram reptile can provide o.08ml of blood = enough blood to fill # 8 microhematocrit tubes. This can provide invaluable information such as hematocrit, total solids, icteric index, and selected biochemistry tests. And, from a smear made on a coverslip, one can determine thrombocyte estimate, white blood cell (WBC) estimate (# of WBC's in 10 field at 40X divided by 10 and multiplied by 2,000), and the presence of intracellular and / or extracellular parasites. However keep in mind that heparin creates a blue tinge to blood films and causes clumping of the thrombocytes and leukocytes. Blood that contains no anticoagulant provides the best samples for evaluating stained blood smears during hematologic evaluation. Remember to always properly label and save any extra blood samples in the refrigerator for possible further testing or rechecking.

Amphibians

Blood sampling in amphibians presents unique problems to the veterinary clinician with the most accessible sites being the lingual plexus, ventral midline sinus and the heart. Unless a blood culture is to be performed on the sample, it is important to heparinize the syringe in venipuncture attempts to avoid coagulation of the sample before it is transferred to the appropriate whole blood tube. If a blood sample is to be obtained for microbial culture, no anticoagulant should be used to pre-treat the syringe, needle, or capillary tube, as this might affect the growth of some organisms. Lithium heparin is the anticoagulant of choice for pre-treating the syringe and for storage of blood since it does not affect the values of plasma calcium, sodium or ammonia. The use of transillumination, along with possible required while often requiring sedation, makes cardiac puncture easier and reduces the risks associated with "blind" venipuncture.

Snakes

Cardiocentesis can be used for blood collection in all snakes. The heart is the first palpable mass located approximately 1/3 the distance from the head. Though its location may vary significantly in certain species. Often, cardiac contractions can be visualized when the snake is placed in dorsal recumbancy. Using one's index finger and thumb to apply gentle pressure, the hearts exact location is isolated and stabilized between these fingers. After disinfection of the ventral scutes in this area, a 22.0 or 25.0 guage needle attached to either a tuberculin or 3.0 ml syringe is slowly inserted at a 45 degree angle into the ventricle and with gentle aspiration the blood may be withdrawn. If the aspirant appears transparent, pericardial fluid has probably been collected. The needle should be withdrawn and another puncture may be performed using a new needle &and syringe. In larger snakes, an alternative site for blood collection is the ventral coccygeal (tail) vein though if venipuncture is attempted too close to the vent one runs the risk of puncturing either the hemipenes or musk glands. Other sites have been described but are difficult to access
i.e. palatine vein, orbital plexus.

Lizards

The ventral coccygeal (tail) vein is most commonly used in lizards, though care should be exercised in lizard species that undergo tail autotomy. The venipuncture site is approximately ¼ the length of the tail caudal to the vent. As in snakes, if venipuncture is attempted too close to the vent one runs the risk of puncturing either the hemipenes or musk glands. Care must be taken to use a long enough needle to be able to reach the ventral vertebral processes. The needle is inserted between scales at approximately 90 degrees into the midline on the ventral aspect of the tail until it comes into contact with a ventral vertebral process. I begin gentle aspiration as the needle is being advanced and often I am able one to collect the sample before the needle reaches the vertebral process. If the needle reaches the process then it is slowly withdrawn the needle 1.0 – 3.0 mm and gently aspirated to collect the sample. An alternative approach to reach the ventral coccygeal vein it a lateral approach. For this the needle is inserted from the lateral aspect of the tail between scales and between the two large muscle bodies in the middle of the crease of the tail. Again the goal is to reach the ventral coccygeal vein. When the ventral tail vein is not productive, the ventral abdominal vein may be used. It is suspended in a broad ligament 1-2 mm within the coelomic cavity on the ventral midline between the umbillical scar and the pelvic inlet. Again caution must be exercised for this is a "blind" venipuncture - and the potential for internal/intra-coelomic hemorrhage exists. Less commonly used is the brachial plexus which is immediately caudal to the elbow joint. In diminutive lizards cardiocentesis via the aid of a Doppler may be required.

Chelonians

The preferred place to collect blood from turtles &and tortoises are the jugular vein because these veins have minimal collateral lymphatic vessels. In many specimens the right vein is usually larger. Each jugular vein typically runs perpendicular and caudal to the ipsilateral tympanum. Sometimes the jugular veins can be visualized when digital pressure is placed on the thorasic inlet. In some chelonians (i.e. marine turtles) there is a postoccipital vein or plexus that arises from the right jugular vein and is located between the occipital protuberance and the cranial border of the carapace beneath the ligamenta nuchae. A brachial vein or plexus is located behind the elbow joint under the prominent/palpable tendon of the insertion of the biceps brachii. With both these venipuncture sites - the access is "blind" - and the samples may be contaminated with lymph altering the biochemical parameters. Lymph when mixed with whole blood dramatically decreases the white cell count, hematocrit, total solids, sodium, potassium, and chloride values. Chelonians also have a dorsal midline tail vein that is best approached at the base of the tail. Cardiocentesis may also be performed but requires a hole be created in the plastron through which a needle may be introduced.

Proctodeum/urodeum/coprodeum - fecal sample collection

Collecting a fecal sample in a reptile/amphibian can be problematic - most reptiles/amphibians defecate infrequently - especially if they are ill and/or anorexic. However, evaluation of the feces for protozoal, amoebic, nematode, etc... Parasites should be considered an essential part of the minimum database for Herp. Patients. Some species do defecate daily, and a sample can be collected at the next voiding. Or in some cases, placing the animal in warm water shallow enough for them to keep their head out will stimulate defecation. In large enough patients – a gloved digital cloacal exam is performed though not usually in the presence of the owner. This procedure may also stimulate the production of cloacal contents. If no sample can be passively collected - then a colonic wash can be performed. The Proctodeum/Urodeum/Coprodeum are quite distensible and the goal is to collect fecal material - so a large gauge tube is used. The tube is properly lubricated and inserted into the colon area through the vent. In snakes care must be taken for the colon is located at the ventral most aspect of the cloaca, and if the tube is directed dorsally - it will enter a blind pocket. Sterile saline 10 ml/kg is typically injected into the colon - and the coelomic cavity is gently massaged prior to aspiration of the sample. In some instances, this procedure will stimulate voiding and a larger sample may be obtained. The sample should be evaluated by culture &and sensitivity (if indicated), fecal flotation for ova, and a direct wet mount for protozoa i.e amoebas and flagellates.

Urinalysis228

Urinalysis in reptiles and amphibians and amphibians can be a useful, economical, and rapid diagnostic tool in assessing the health status of many specimens. Because there are significant differences between reptile and mammalian urine, the interpretation is quite different. In reptiles, uric acid is the principle product of protein catabolism instead of urea. Precipitates of uric acid are normally passed as hard concretions. Reptilian kidneys lack loops of Henle, which means that reptiles consistently produce isosthenuric urine with a specific gravity of 1.005-1.010 regardless of their hydration status. Reptile ureters empty into the cloaca. Urine is subsequently either stored in the cloaca (snakes do not possess urinary bladders) or refluxed into a bladder (some lizards and all chelonians). This means that microbiological culturing of urine (even by cystocentesis) can lead to erroneous interpretation. The color of reptile urine varies from transparent to brown depending upon the diet and the presence of bile pigments. Although renal thresholds for blood glucose have not been evaluated in reptiles, glucosuria is abnomal and has been observed in cases of diabetes mellitus. Hyperglycemia has also been found to be associated with inflammatory diseases of the coelomic cavity, hepatic lipidosis, and severe wasting in cheloneans. It is reported that in these instances, it can be transient, resolving without insulin therapy – though it may also indicate a grave prognosis. Also when evaluating for glucose in the urine, it must be remembered that the urine glucose reagent pad can show false positive results from contamination with substances such as peroxide and hypochlorite. False negative results may occur if the urine is refrigerated, or contains formaldehyde or vitamin C. Ketones do not appear in the urine of hyperglycemic, glucosuric chelonians. Though protein may be present in small amounts in the urine of "healthy" domestic mammals, "healthy" chelonians appear to have only trace amounts of protein in the urine. However because the protein assay is sensitive to a variety of other factors it must be interpreted with care. Contamination with fecal matter, semen, or egg material could introduce proteinaceous materials into the urine thus rendering a false positive result. Also a high urine pH can cause a false positive result by interference with the buffering mechanism on the test pad. If all these factors are taken into consideration and ruled out, then elevations in urine protein levels may indicate glomerular damage and plasma protein leakage into the urine.

Repeated retesting is indicated, and consistently high proteinuria along with other signs of kidney disease would point towards the need for further renal evaluation via biochemical testing or biopsies to accurately diagnose the disease process. Urine pH can be influenced by a variety of factors including diet, mixing with fecal matter, nutritional status, metabolic or respiratory acidosis, and possibly urinary tract infection with urease-producing bacteria. Herbivorous reptiles normally produce alkaline urine with a pH of 7-8. Carnivorous reptiles typically produce urine with a pH of 6-7. Anorexic tortoises in a state of metabolic acidosis and those emerging from hibernation can produce acidic urine. Microscopic analysis is without doubt one of the most important aspects of the urinalysis. The presence of red blood cells and white blood cells is abnormal and can indicate infections of the lower urinary or gastrointestinal tract, or possible cystic calculi. The presence of parasites in the urine indicates the need for proper treatment using appropriate parasiticides. It has been reported that urate crystals can take many forms, and could be confused with cellular casts. However, when a large number of waxy and/or other cellular casts are found – this is a definite sign that further diagnostic testing is indicated including plasma biochemical analysis, radiography, and possibly laparoscopy with renal biopsy.

Trans - tracheal sample collection

Trans-tracheal collection of material is indicated for best evaluation of respiratory disease in reptiles & amphibians. The sample should be evaluated by aerobic culture &and sensitivity testing, direct wet mount, and cytologic examination. Lung worm (Order: Rhabditida) can be a serious problem in reptiles & amphibians, and the parasite ova may be observed on a wet mount sample.

The lungs of most reptiles are simple, sac like structures with ridges for increased gas exchange. They do not have alveoli or bronchioles. Because they only have a trachea and two main stem bronchi which open into the lung, many samples collected from the trachea are actually lung washes. Frequently sedation is not required in debilitated animals. Appropriately sized sterile feeding or urethral catheters work well. Sterile saline at 5.0 – 10.0 ml/kg is instilled into the trachea/lung after passing the catheter through the glottis. In chelonians and lizards with unilateral pulmonary lesions, the tip of the catheter has to be bent to ensure that the catheter enters the affected lung. Snakes w. Which have one functional lung, can have the catheter passed straight down the trachea into the lung. After the sterile saline has been instilled into the lung, the patient is gently rotated to loosen any exudate. Aspiration is then performed, and harvested material examined using cytology, parasitology and bacterial culture and sensitivity.

Concerning culture and sensitivity the most important thing to assure the most successful outcome of culturing pathologic microorganisms is to properly handle the specimen(s) collected. It is important to contact your specific laboratory to determine their exact requirements and recommendations for submitting samples.

Radiography

Radiographs are often indicated as part of a complete work-up and are an invaluable diagnostic tool for identifying numerous diseases in reptiles. Some indications for radiographing specimens are to aid in diagnosing "metabolic bone disease", skeletal fractures, osteomyelitis, articular gout, soft tissue mineralization, lower respiratory tract disease, foreign bodies, constipation, gravidity, dystocia, coelomic masses, coelomic effusions, hepato &and renomagaly, and cystic calculi. The exoskeleton of chelonians, thick dermis of many lizards, and strong muscle tone of snakes make diagnosing these and other disorders via palpation during physical examination challenging. The settings for most radiographs are determined on a case by case basis. Ideally one should use the high-detail, rare-earth intensifying screens and compatible film for reptile and amphibian radiography. With the increasing popularity and benefits of digital radiography – this is the wave of the future and so equipment and techniques will change. However, none the less for all reptiles & amphibians it is important to develop and maintain a proper technique chart for quick reference when taking radiographs.

If possible, a horizontal beam should be used for lateral and craniocaudal (cc) views, especially in chelonians. To be able to produce a horizontal beam x-ray exposure the x-ray tube has to have the capability of being rotated 90 degrees so that the x-ray beam can be directed horizontally across the table rather than down from above towards the table. It is still recommended that the distance between the x-ray tube and the x-ray cassette (focal film distance) still be 40 inches, as is the case for most vertical Table Top radiographs. Horizontal views permit organs to stay in their normal positions and prevent artifacts. This is especially important when imaging lung fields due to the fact that reptiles and amphibians do not possess a diaphragm that would separate the chest cavity from the abdominal viscera. This will allow the visualization of the right and left lung fields as separate structures. In larger snakes and lizards, numbered identification markers should be placed on sections of the body to better determine the exact location of lesions and organs. It is recommended that the veterinarian maintain a set of normal radiographs for each species, which can be easily retrieved and compared to those of diseased animals.

Ultrasonography

Ultrasonography is a very useful tool in reptile &and amphibian medicine. Because of their bony shell - imaging in chelonians can only be performed through the flank and axillary areas. The organs in the coelomic cavity can be visualized and differentiated by their specific ultrasonographic texture. The ultrasound can then be used to image and guide the biopsy of the various organs including the liver, spleen, cystic structures, kidneys, et cetera. As always, it is very important to know the normal anatomy of the species when interpreting the image produced by the ultrasound.

Endoscopy

Endoscopy has proven to be a most useful diagnostic tool in veterinary medicine. Specifically in the field of zoological medicine, the application of diagnostic endoscopy has shown great promise in a variety of species and has been used extensively in avian medicine &and surgery23surgery9. Flexibe &and rigid endoscopes are being used for a number of different applications in reptile &and amphibian medicine &and surgery i.e. coelioscopy, pneumoscopy, gastroscopy, + direct visual biopsy.

Computorized tomography (CT)

This is an excellent though not always available means to evaluate bony lesions. Computorized Tomographic scanning produces transverse x-ray slices (tomographs) of tissue and is particularly sensitive to calcium and/or air containing structures.

Magnetic resonance imaging (MRI)

This too is an excellent though not always available means of evaluating soft tissue lesions. The magnetic resonance imaging utilizes tissues magnetic properties and response to radio wave pulses - producing high detail images of all tissues except bone.

With the continued advancement and popularity of these last two diagnostic modalities in human medicine – both the CT as well as the MRI are becoming increasingly more available to veterinarians and the non-invasive benefits of being able to evaluate exquisite detail in multiple dimensions is making these procedures more and more appealing.

Review of therapeutic techniques in reptiles & amphibians

Sites &and techniques for injections

Injections may be given intravenously, subcutaneously, intramuscularly, intracoelomicly, intracardiacly or intraosseously.

Intravenous injections are given in the sites described for blood collection. Because of the potential involvement of the renal portal system, it is recommended by some by many that most injections be administered in the cranial half of the body. One exception to this is intracoelomic injections -which are typically given in lizards in the lateral right caudal quadrant region at a level even with the cranial aspect of the rear leg. Gentle aspiration of the syringe assures that the needle is not in any vital organ. In chelonians the extremities may be used for intramuscular (i.m.) injections of small volumes. For larger volumes - the pectoral muscles are the preferred site for i.m. injection. These large muscles attach the front limbs to the plastron. The injection is given by inserting the needle parallel to the plastron under the arm. Intracoelomic injections are given in the flank near the junction of the skin with the shell just cranial to the rear leg. Again gentle aspiration prior to injection assures that the needle is not in any vital organ. Intracardiac injections are generally reserved for situations when no other vascular access is available.

Sites &and Techniques for Catheter Placement

Amphibians

Intravenous catheterization is not easy nor well tolerated.

Snakes

The jugular veins and the heart are at present the only accepted sites for catheter placement. The right jugular vein is typically larger that the left and is therefore the best choice for catherization. Catheter placement requires a cut-down incision be made from approximately the 4th to the 7th scute cranial to the heart at the junction of the ventral scutes &and the right lateral body scales. The catheter is introduced cranial to caudal. Cardiac catheterization is used in snakes for short term, up to 24 hours, or emergency procedures. The technique is the same as that used for cardiocentesis. There are no accessible intraosseous sites in snakes.

Lizards

The cephalic veins are the preferred site for IV catheterization. The dorsal (anterior) surface of the antebrachium is prepped for a sterile procedure - cut-down and dissection of the vein is required in most cases. The skin incision should extend from the dorsal proximal limit of the antebrachium medially to allow the best visualization of the vein24vein10. Intraosseous catheters may be placed in most lizards. The author prefers the proximal tibia. Insertion of the catheter into the distal femur has been reported, however - presently it is recommended that this location should be avoided as the stifle joint is invaded and the intercondylar cartilage is damaged25damaged11.

The proximal tibia is easily accessible and insertion through the craniomedial aspect of the bone avoids invasion of the joint capsule and articular cartilage. A spinal needle (size &and length determined by the size of patient) is used and the needle catheter is drilled into the tibia while palpating the direction of the bone. The placement can be tested by aspirating and obtaining a small flash of blood. If no flash back is observed, a small amount of saline is injected and the aspiration is attempted again. If the catheter is outside the bone, the muscle will swell with the injection of saline. Ultimately, radiographs can be used to confirm the location of catheter placement25placement11.

Chelonians

As with venipuncture, because the right jugular vein is larger than the left in many species, it is the preferred site for catheter placement in turtles &and tortoises. Animal with thick skin over the jugular vein or those with poor peripheral blood pressure may require cutdown &and dissection to identify &and catheterize the vessel. There is continued discussion as to whether Cchelonians do not have marrow containing long bones25bones11. However, it has been reported that an intraosseous catheter can be placed in the cranial or caudal aspect of the bony bridge - the column of bone between the plastron &and carapace24carapace10.

Sites &and techniques for fluid therapy

Signs of dehydration in reptiles & amphibians can be similar to those seen in mammals. Loss of skin elasticity &and wrinkled appearance progressing to sunken eyes, dry tacky mucus membranes, et cetera - all must be interpreted with the understanding for normal appearances for that particular species. Packed cell volume (PCV) &and total solids can also be helpful in evaluating dehydration.

Placing the patient in shallow warm water usually allows for some absorption of fluids, moisturizes the skin, and in some instances stimulates defecation. Fluid replacement can be orally in mildly dehydrated, alert patients. Subcutaneous fluids are useful in mild to moderately dehydrated patients and patients that will not allow oral access. More rapid routes for fluid delivery for patients with severe dehydration or shock-type situation include intracoelomically &and Intravenously. With repeated i.c.o. fluids it is important to aspirate not only to assure the needle is not in a vital organ, but also to assess that the previously administered fluids have been absorbed.

Significance of &and techniques for thermal support

In that reptiles & amphibians are poikilothermic, thermal support is vital for optimal maintenance of our patients. As mentioned earlier, not providing the proper Preferred Optimal Temperature Zone (POTZ) is often one of the underlying causes for illness. Providing the POTZ is vital to allow for the proper functioning of the immune system, gastrointestinal system, and potentially all body functions. It is therefore very important that the preferred optimal temperature zone for each individual species be known to avoid thermal stress. If this information is not available, most debilitated reptiles will respond to a temperature range around 32.2 C (90 F), until the species specific information can be obtained. Properly used radiant heat sources tend to be best at providing a safe temperature gradient. Incubators can be used, however they tend to provide a relatively constant temperature - generally adequate only for short term maintenance. Typically I do not recommend “Hot Rock” type heat emitters for they can at some time fail and can often therefore result in different degrees of significant thermal injuries.

Significance of and techniques for delivering nutritional support

As mentioned many reptiles & amphibians presenting are malnourished either because of dietary deficiencies or improper husbandry having an adverse effect on the animals metabolism. It is generally recommended that nutritional support be provided to any patient that has acutely lost 10% of chronically 20% of its' body weight. Nutritional support can be provided by a number of means including syringe/force feeding, orogastric tube feeding, and pharyngostomy feeding tube placement. Though at times the greatest challenge is often opening the mouth - once it is open - the glottis, which is typically closed at rest, is located at the base of the tongue in the rostral oral cavity making it easy to avoid during orogastric intubation.

When continued nutritional support is required for reptiles with mouths that are difficult to open, placement of a pharyngostomy tube is recommended. To do this, the animal is sedated, and a mosquito hemostat is inserted into the pharynx and pointed laterally against the wall of the esophagus. The tip of the hemostat is palpated externally, and a small nick incision is made over the tip. The tip of the hemostat is then gently/bluntly dissected out through the hole in the skin. Careful dissection is used for too large a hole will allow reflux of material out the stoma.

A Sovereign (Sherwood Medical, St Louis, MO) red rubber feeding tube (size [Fr. MM] &and length {"/cm} dependent on animal size) is then measured &and marked - so as to have the tip end up in the stomach. The hemostat is then used to grab the tip/gastric end of the feeding tube and the tube is pulled through the hole into the esophagus &and out of the mouth. At this point the blunt/solid tip of the feeding tube is trimmed to open the end, and the tube is then redirected down the esophagus and into the stomach. The tube is then sutured in place using the "chinese finger-trap" technique. It is important to flush the tube after feedings and best to place some sort of cap to close off the exposed end.

For herbivorous and omnivorous reptiles and amphibians, Isocal &and Sustacal (Mead Johnson Nutrionals, Evansville, IN), Ensure &and Osmolite (Ross Labs, Columbus, OH), are appropriate liquid diets. For carnivorous reptiles & amphibians Traumacal ead Johnson Nutrionals), Pulmocare (Ross Labs), and Feline and canine Clini care Liquid (Pet-AG, Elgin, IL) are appropriate liquid diets. Care must be exercised in patients suffering chronic starvation. "Refeeding syndrome", a condition referring to hypophosphatemia and hypokalemia, is seen in patients fed large amounts of calorie rich foods and it can be potentially fatal. The phosphorus and potassium move into the cells with glucose and thereby deplete circulating levels - resulting in rapid weakness and often coma - leading ultimately/often to death. It is therefore recommended that reptiles & amphibians that have been chronically starved be fed 50% of their need calculated based on their real, not ideal weight. This would be continued for several days, until the patient's condition improves. The amount of calories supplemented is then increased in increments of 10 - 20 % until the recommended level of caloric supplementation for that sized animal is reached25reached11.

For an excellent current reference on feeding carnivorous, omnivorous &and herbivorous reptiles please refer to Dr. Scott Stahl’s Proceedings notes and his references for the 2000 meeting of the Association of Reptilian & Amphibian Veterinarians (ARAV), Reno, NV. [pages: 177 – 182].

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