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The Reptile Survival Guide Online 'Mini Series'

Session 1: Critical Care of the Sick Reptile

Kevin Eatwell
BVSc (Hons) DZooMed (Reptilian) Dip ECZM (Herp) MRCVS
RCVS & ECZM Diplomate in Zoological Medicine
RCVS & ECZM Recognised Specialist in Zoo & Wildlife Medicine

REPTILE CRITICAL CARE

Reptiles have a low metabolic rate and so many disease processes progress over time. However many clinical signs are subtle and many cases will present with acute signs of chronic disease and hence will have become emergency cases. Many of these can be related directly or indirectly to inappropriate husbandry and owners must be committed to correcting husbandry errors long term before their pet is returned to them. There is no point in treating a reptile for it to be returned to sub optimal husbandry as it will never recover. As a result we need to provide for the basic needs of reptiles in the clinic as well. Reptiles therefore need secure and escape proof accommodation with suitable heat and light sources. This is short term accommodation so fancy environmentally enriched enclosures are not required.

Lizards should be supported in ventral recumbency, if collapsed or in respiratory distress. Towels or bean bags can be used to elevate the head and cranial thorax slightly to prevent the abdominal contents from placing excess pressure on the lungs. It is difficult to encourage a snake or chelonian to stay in a suitable position. Snakes can classically display orthopnoea when in respiratory compromise. Taking the body weight is important to act as a bench mark for evaluating recovery and drug therapy.

Thermal therapy

Thermal therapy is of extreme importance and knowing the optimal temperatures are critical.

Many reptiles presented can be in a state of collapse. It is impossible to be able to properly clinically assess a cold reptile. So initial supportive care can simply include warming the reptile. Many species we deal with have specific husbandry requirements but most will benefit from being kept at a temperature in the high 20’s or low 30’s. A collapsed reptile is unable to thermoregulate as this requires the ability to move. So we have to place the animal in an even temperature that is appropriate. Hot basking sites are not indicated here. The easiest and most practical solution is to use a heated plant propagator. This is cheep to acquire and run but keeps a nice stable temperature of approximately 28^{0}C. These can also be used to create a high humidity environment. Water bowls placed inside can help elevate relative humidity further. A cold reptile has a lower metabolism and will be unable to absorb food, fluids or any drugs given. Wait until the reptile is warm before providing any supportive care measures or drug therapy. Vivaria are more suitable for hospitalisation of any length of time for lizards and snakes. Heat sources used in these environments can include heat mats and ceramic spot bulbs. Tortoises are best hospitalised in open top boxes, for larger species they will need to be of sturdy construction. Heat and light should be provided from overhead and not underneath. All heat sources should have their temperature monitored and controlled. The surface temperature of larger reptiles can be monitored by attaching a small spring dial thermometer.

Fluid therapy

Providing a water source in an appropriate fashion is important! Reptiles drink by a variety of methods; ranging from submerging their entire head underwater (chelonians) through to licking droplets from foliage.

Many reptiles will be presented by the owners after a long illness. This means that many critical patients may be exhibiting acute clinical signs resulting from a chronic illness. Many will have been kept in sub optimal environments. It is therefore likely that many will be dehydrated. However given the wide range of compensatory mechanisms that they possess this may not be clinically evident as reptiles are able to preserve plasma volume despite being dehydrated. Once there are clinical signs of dehydration the situation has exceeded all the compensatory mechanisms discussed.

In many cases with reptiles dehydration must be assumed in the first instance. With a priority placed upon quantifying dehydration. All reptiles are capable of changing their plasma osmolarity based on the availability of water in their environment. Terrestrial chelonians for example can have a blood osmolarity that varies between 250mOsmoles and 450 mOsmoles. Lizards also vary blood osmolarity based on water availability. So blood osmolarity can be a useful guide to the hydration status. Terrestrial chelonians use plasma urea to elevate blood osmolarity as this molecule is easily diffusible across membranes and easy to excrete when water is available. Well hydrated chelonians have values of less than 2.1mmol/L. Dehydrated individuals can have values in excess of 40mmol/L. The level of dehydration can also be assessed in all reptile species by checking the plasma sodium levels.

In chelonians assessing urine is a useful technique for assessing the hydration status and the urine specific gravity and pH can be used. As the animal becomes dehydrated the urine becomes acidic (for herbivores it decreases below the typical pH of 7.5) and the specific gravity increases (greater than 1.012 is considered to be consistent with dehydration). Ketogenesis within the reptile would lead to ketones being present in the urine.

In all species the urine output (volume and frequency) is a good guide to hydration levels, provided the animal has not been scared as this will encourage it to void the contents, as a reptile will only void urine when it has sufficient fluid on board.

Measuring body weight is a useful parameter as it gives a guide to the successful rehydration of a reptile. Weights recorded must be done so at the same time each day.

The aim of hydration is to restore the plasma to the anticipated osmolarity of the species given its physiological status and the time of year.

The first step in rehydration of a reptile is to get the relative humidity right. Low humidity can lead to a number of clinical signs, which are not covered here. Relative humidity can be increased by providing water bowls placed on heat mats, dripping, misting or fogging systems within the vivarium. Waterfalls or damp substrate can also be utilised. A hand held water sprayer aimed at the reptile or misted into the enclosure is helpful. Reducing ventilation in the enclosure in order to raise humidity is not acceptable.

Once the reptile has warmed up, fluids therapy can be provided. Reptiles have a maintenance requirement of 30mls/kilo/day. Once again a variety of routes can be utilised.

Bathing (under supervision) is an excellent non-intrusive, atraumatic method for hydrating snakes, lizards and chelonia. This water should be 25^{0}C temperature. Warm water bathing also serves as a stimulus for voiding urates and faecal material. The passing of urine is a sign that the reptile has rehydrated itself as a reptile would not inadvertently waste it water store. In a sense bathing allows dialysis of a reptile via the bladder. Some snakes and lizards may be initially reluctant so it is advisable to use a bathing container with a lid. Supplements can also be added in the form of electrolytes. A warm bath for twenty minutes twice a day can dramatically improve the fluid balance of many reptiles. This is partly due to the fact that they may drink from the bath, and also for chelonia and some lizard species, due to fact they can take up fluids via the cloaca into a bladder. Snakes and some other lizard species can also take up fluids into the cloaca from where it can pass to the caudal part of the intestine. The reptile bladder differs from that of mammals in that it is not directly connected to the urinary system. The bladder joins onto the cloaca and acts as a storage area for urine and fluids which can be absorbed or flushed into the large intestine for absorption. Weighing a reptile both before and after a bath is a good way of assessing the degree of rehydration that has occurred and is a useful technique to convince owners of its importance. If the reptile is critically ill bathing could lead to drowning and it is highly unlikely the reptile will voluntarily take water in. Small or docile reptiles can be syringe fed into the mouth. If this is not appropriate, then a suitably sized tube can passed into the stomach. 1% of bodyweight can be given by stomach tube. However a bevery ill reptile will be unable to deal with enteral fluids and there is a risk of poor assimilation or regurgitation. Many authors consider rehydrating a reptile at a rate of 4% bodyweight per day.

Chelonia have a powerful bite and a sharp beak, so a gag and a metal tube may be required, particularly for larger specimens. There are also reports of tortoises ingesting feeding tubes. Always use some form of gag and a leur fitting syringe. Lizards possess teeth that could be easily damaged. Jaw fractures have also occurred as a result of tubing with metal tubes. Larger snakes will require fairly long tubes. For these species metal tubes must be used with caution. However, cat and dog urinary catheters make ideal atraumatic feeding tubes. Wooden tongue depressors and plastic needle covers make suitable gags since they are also atraumatic. Before passing the tube it is advisable to measure how far it will need to pass into the animal in order to reach the stomach, and a mark made on the tube. This will be about 50% of the snout to vent length in snakes, the rib cage in lizards and for chelonia down to the junction between the pectoral and abdominal plastron scutes. The tube should be lubricated before being introduced either centrally, for example snakes, or from the patients left to right, over the back of the tongue, depending on the most suitable approach for the oral anatomy of the reptile.

Injectable fluids are therefore the preferred route and can be given intracoelomically, intra osseously, intravenously or epicoelomically (chelonians). In most situations the intraosseous, epicoelomic or intracoelomic routes are the easiest to perform.

Parenteral fluids can be given at the rate of 1% of bodyweight up to four times daily. Many authors consider rehydrating a reptile at this rate until rehydration has been successful (usually judged by voiding urine). Fluids used for mammalian species are acceptable as the sodium content and the osmolarity of these fluid matches the levels found in reptiles. Studies have also shown that reptiles have a larger intracellular component and using lower osmolarity fluids have been proposed. Given current information, diluting fluids with an additional 10% of sterile water for injection, as was previously reported, is unnecessary as mammalian fluids will provide additional water for most sick individuals. Even species with hypotonic plasma (such as fresh water turtles) may well benefit, depending on the case, as fresh water species tend not to dehydrate when ill but may be over hydrated. This is because the plasma pumps, which maintain their hyperosmostic state (relative to fresh water), fail and this is also why hibernating turtles progressively get hypotonic during hibernation). There is some logic in replacing deficits with higher osmolarity fluids (cellular fluid is being replaced) and lowering the osmolarity for maintenance (intravascular fluid is being maintained). All sites should be disinfected with an iodine based scrub prior to injection.

Snakes can be injected subcutaneously with fluids in the caudal third of the dorsum, laterally. In lizards the lateral thoracic area can be used but skin may darken at the site. Chelonia have skin folds cranial to the hind limbs and caudal to the neck area that can be used for subcutaneous fluid administration.

In chelonians epicoelomic fluids can be given just above the plastron in between the head and foreleg. This fluid is delivered into the potential space between the pectoral muscles and the plastron. It is considered that there is a good degree of vascularisation in this region as the pericardial fluid in chelonia can act as a fluid store during periods of drought or hibernation. This is a simple technique that can be done single handed by the veterinary nurse.

Fluids can also be given intracoelomically to all species. In chelonians the prefemoral fossa is the best site. The tortoise is tipped away from the handler and the injection delivered into the lower section to avoid injection into the lungs. In lizards the best site to inject is into the caudal quarter of the coelomic cavity off the midline on the ventral aspect with the lizard turned over. Care has to be taken to avoid the lung fields and this is of particular important in the chameleons as they have finger like projections from the back of the lung (dendritic processes). In snakes injecting into the last quadrant is best. Draw back on the syringe prior to injection to check the viscera have not been punctured. The down side of this route is slow absorption and this can interfere with subsequent diagnostics such as endoscopic examination and lead to increased intracoelomic pressure and lung compression due to sequential overloading as repeat doses are given. Many authors recommend withdrawing on the syringe before repeat doses. If fluid is aspirated there is no point injecting further fluid into the cavity. As the tortoise is tipped away from the handler the risk of puncturing the bladder is minimised. I would not use this route for repeat administration in severely debilitated species as coelomic ascites is not an uncommon clinical presentation. Getting the fluids into the circulation is what is important in these cases not just into the patient. I would only consider this technique as a one off procedure in animals that do not have ascites and it could again be performed by the veterinary nurse when the animal is admitted. For chelonians I would prefer the epicoelomic route.

Intraosseous techniques are ideal for limbed lizards. This however is generally a two person job and should involve the veterinary surgeon. Many sites can be used but I prefer the tibia. The limb is scrubbed clean using iodine and then surgical spirit is applied. Local anaesthetic is injected over the proximal end of the tibia. The maximum dose of lidocaine is 4mg/kg to avoid toxicity. A spinal needle is inserted down the full length of the medullary cavity. Another popular choice is the proximal femur. Here the concavity between the trochanter and neck is palpated and the needle inserted down the shaft of the bone. Intraosseous techniques can be difficult in tortoises as they have limited areas of medullary bone. The classical site reported is in the bony bridge between the plastron and carapace. A drill is required to gain access and a spinal needle placed into the hole and taped in place. It is easy to misplace these and some end up being actually intracoelomic needles. The tortoise should also be anaesthetised before drilling. A novel site has been recommended using the plastron between the head and forelegs. This site would not lead to incorrect placement. Intraossesous techniques are not practical in snakes. Reptiles cope well with an attached syringe driver and this can be taped to the chelonian shell.

Intravenous fluids are an option as well but are more technically demanding. In chelonians the right jugular vein is used (as it is larger than the left). This runs dorsally from the tympanic membrane towards the base of the neck and a 24 gauge catheter can be sutured in place. In lizards the cephalic or jugular vessels can be used. The jugular vein courses from the point of the shoulder to the angle of the jaw. Typically a surgical cut down is required to suture a catheter in place. The cephalic vein courses across the radius and ulna and again a surgical cut down is required. In snakes the jugular/azygous vein can be used. The apex beat is identified and a surgical incision made 12 ventral scales cranial to the heart on the right hand side just below the end of the ribs. The incision is made between the second and third row of scales. The jugular vessel lies just at this point and a catheter can be sutured in place. Intracardiac catheters have also been described but haemopericardium is a potential complication leading to cardiac tamponade. In lizards using the tail vein for catheterisation has also been described but in this authors personal experience has commonly become displaced and fluids going perivascular. The ventral abdominal vein can also be used for catheterisation in lizards. Sadly in extremely collapsed individuals vessels may be difficult to locate for catheter placement. Fluids could be given by injection via other routes to improve perfusion prior to catheter placement.

Crystalloids for replacement therapy include lactated ringers solution. There have been some concerns regarding the use of lactate in reptiles as they may exacerbate the lactic acid levels in the blood. Lactate is converted by the liver to bicarbonate to buffer the solution. So lactate provided by fluid therapy does not increase plasma lactate levels, unless there is end stage liver failure and the liver is unable to convert the lactate. These fluids should be warmed to body temperature prior to injection.

Colloids can also be given to reptiles to maintain the colloidal osmotic pressure within the circulation. These replace the intravascular fluid and must be administered by intravenous or intraosseous routes only. They draw fluid into the circulation and must be given with crystalloids to prevent interstitial dehydration. It has been suggested that they are administered 50:50 with crystalloids. Blood transfusions have also been reported but there is little information available on suggested techniques. Once again they must be given via the intravenous or intraosseous routes only.

Measuring body weight is a useful parameter as it gives a guide to the successful rehydration of a reptile. Weights recorded must be done so at the same time each day, it is in fact useful to weigh hospitalised reptiles at least twice a day. Initially weight would be expected to increase dramatically as the dehydrated anorexia reptile becomes full. Thereafter weight will stabilise.

Supportive nutrition

Reptiles are designed to conserve water and as a result many animals presented can be electrolyte deficient despite having reasonable plasma values. This is because there is cellular depletion of electrolytes. Providing a sudden bolus of glucose to a reptile can lead to a significant drop in plasma electrolytes and it is important to ensure the reptile is hydrated well and provided with electrolytes prior to supportive nutrition. If the reptile is looking brighter or has passed urine, stomach tubing and bathing should be added to the regime and parenteral fluid therapy can cease. The metabolic rate of reptiles is between 2 – 5% of a comparably sized mammal thus there is no urgency for nutritional support and rehydrating the patient first is a simple rule of thumb.

Commercially available preparations are available for tube feeding and a reptiles' stomach can take up to 1% bodyweight volume at a time. Very weakened animals will require less than this.

It is advisable to over dilute oral electrolytes for reptiles, this prevents concentrations being higher in the gut than the ECF and causing water to move from the body into the gut. The preferred initial formula to use is critical care formula® (Vetark). This provides energy in the form of carbohydrates and protein but no fibre. Recommended dosage is 1 scoop per kilo bodyweight per day. Subsequently herbivores can be given critical care for herbivores® (Oxbow) can be used, which can be mixed with pureed vegetables/fruit as required. In carnivorous species, despite concerns regarding its purine content, A/D® diet (Hills) is used frequently. This is a problem as the purines are metabolised directly to uric acid and can lead to a potentially increased risk of gout formation. More recently critical care for carnivores has become available and this has a higher pyramidine component. In either case waiting for the animal to be successfully hydrated first is a wise move. This can be diluted with warm water or critical care formula as required. Snakes do potentially present some concerns when supportive feeding as naturally they undergo fasting for some time and then elevate their metabolic rate and intestinal volume. By 48 hours after feeding, the metabolic rate of the animal can elevate 8 fold and the intestinal mass doubles. This involves a huge energetic outlay and you need to make sure sufficient food is available to justify this. It may seem wise to tube a snake until it is fit to burst then leave it alone, however, practically frequent tube feedings have not been reported to be detrimental to snakes. It is difficult to quantify the precise metabolic requirements of an individual in the clinic and it is perhaps practically easiest to monitor body weight change over time. For more long term nutritional and fluid support an oesophagostomy tube will need to be placed.

Oesophagostomy tube placement

For longer term nutritional support an oesophagostomy tube is required. This will briefly be covered as it is an important aspect of supportive care. Oesophagostomy tube placement allows for continued fluid and nutritional support and a route for oral drug therapy. These are most commonly used in chelonians but are equally applicable to squamates. The key to keeping an O-tube in place in chelonians is to situate the tube far enough back so that the tortoise is unable to hook out the tube with its leg. The tube can be placed on the left or the right side. Many authors consider placement on the left best as the tortoise has a larger jugular on the right side and the oesophagus curves to the left. Practically it is easier to place the tubes on the right side if you are right handed. A pair of curved haemostats are introduced into the oesophagus and displaced laterally. Care should be taken to avoid the jugular vein and carotid artery. The skin tents and usually the vessels slip dorsally or ventrally. The skin is cut with a scalpel blade and the haemostats pushed through. The feeding tube is grasped and pulled through the incision and out through the mouth. It is best not to cut it to length at this stage (as it is easier to pull through the incision) but measuring and marking the tube before beginning surgery is wise. Once pulled out the mouth the tube is cut to an appropriate length and directed back down the oesophagus.

The tube can be secured using a Chinese finger trap suture or using surgical tape and sutured to the skin with horizontal mattress sutures. In aquatic species securing knots with superglue is advised. Dressing the leg can be useful as it covers the rough scales over the elbow joint further reducing the changes of the tube being pulled out. Duck tape has proved to be consistently the best product to use on leg dressings.

Tortoises are perfectly capable of eating voluntarily even with the tube in place. Once this has occurred and the tortoise is feeding well, supplemental feeding can be stopped. Once the tortoise is holding weight without supportive care the O-tube can be easily removed conscious.

In lizards O-tubes are placed mid way down the neck and secured over the dorsal spine. O-tubes can be placed in snakes and can be secured by suturing to the scales. Any incision should be made between scales. The tube can be secured using surgical tape and sutured to the skin with horizontal mattress sutures. The length of tube should be sufficient to reach the stomach or distal oesophagus in snakes.

Patient monitoring

Once the tube is in place the bodyweight of the patient should be monitored. Supportive nutrition should continue until the weight is stable and the reptile is feeding for itself. Then the supportive nutrition can stop whilst the weight is being monitored. If the weight is stable over 2 – 3 weeks the oesophagostomy tube can be removed and the hole left to granulate.

Emergency Drug therapy

In critically ill patients emergency drug therapy may be required in addition to the basic care outlined above. However it is important to warm any patient prior to any drug therapy.

Oxygen therapy

Respiration in reptiles is stimulated by hypoxia and hypercapnia and placing a reptile in a high concentration oxygen environment may actually inhibit respiration. Many authors consider supplementing room air with oxygen up to a percentage of 40% combined with humidification. There are units available commercially or a nebuliser can be used with an oxygen supply into the vivarium. If ventilation is required then the animal can be intubated and ventilated in oxygen using a mechanical ventilator or by bagging on a t piece otherwise an inspirator or ambu bag can be used to ventilate in room air.

Reptile analgesia

Lesions that are painful in mammals are also likely to cause pain in lower vertebrates. Clinical signs of pain tend to be limited but can include withdrawl into the carapace, restlessness, aggression, stenting of the region in snakes (areas held rigid and no curling) and increased respiratory rate. Quantifying the effectiveness of analgesics can be difficult as a result.

Non steroidal anti-inflammatory drugs should be used as a routine. There has been a pharmacokinetic study using meloxicam in green iguanas. This means this is the agent of choice until further studies are performed. An intravenous dose of 0.2 mg/kg was effective for 48 hours. A single oral dose failed to achieve therapeutic levels. A recent pharmacokinetic study in green iguanas using ketoprofen has been published. A dose of 2 mg/kg intravenously was suggested and achieved therapeutic levels for over 24 hours. Dosing every 36 hours would seem to suffice.

Opioid analgesics are used frequently in reptiles. Butorphanol has been used at 0.02 – 25 mg/kg in the clinical setting. Their effectiveness has been judged by anaesthetic sparing effects. However many reports fail to demonstrate this effect. In green iguanas 2 mg/kg did not alter the heart or respiratory rate, 1 mg/kg did not show any anaesthetic sparing effect and 1 mg/kg did not reduce the amount of isoflurane required to kill them either. The response s of red eared terrapins to a painful stimulus were compared. One group were given 28 mg/kg butorphanol and there was no difference in the withdrawl response. There was respiratory compromise as a result. Clinically many vets feel the animal is less painful as a result of its administration. More recent studies have suggested dosages of 1.5 or 8 mg/kg in green iguanas. Recent studies comparing butorphanol and meloxicam in ball pythons failed to show any reduction in the physiological stress response in animals undergoing surgery. Morphine is generally used in preference at 1 – 2 mg/kg.

Antibiotic therapy

A hypothermic reptile may well have a reduced total white cell count and covering antibiotics are often given as the reptile is immunocompromised. It is important to select an appropriate antibiotic which will cover against likely pathogens. If possible samples for cytology and culture should be taken prior to starting therapy to quantify potential pathogens. It is important to provide broad spectrum cover against the common secondary pathogens in reptiles. These are primarily gram negative rods such as *Pseudomonas*, *Aeromonas* or *Proteus*, however 54% of reptile infections include anaerobes and attention should be paid to providing antibiotic cover for these as well. The only antibiotic licensed for use in reptiles, is enrofloxacin. This is bactericidal and effective against aerobic bacteria and *Mycoplasma sp* only. As an alternative, ceftazidime is a third generation cephalosporin with excellent anti-pseudomonal activity. It is bactericidal and kills aerobic and anaerobic bacteria. There is no reason not to combine both these agents on initial presentation. Drug therapy can be amended based on culture results. Other agents used include anti pseudomnal penicillins such as ticarcillin and aminoglycosides such as amikacin.

Identifying death and resuscitation

Owners may telephone regarding a collapsed, presumed dead, reptile and it is important to persuade them to present this for examination to confirm death. In severely collapsed individuals where the heart rate is slow the use of adrenergic agents is questionable. These lead to a greater left shift in reptiles increasing the bypass of the lungs and do not improve tissue oxygenation. Atropine and glycopyrrolate have been used in reptiles in an attempt to elevate heart rate and it has been shown that they do not. If an animal is presented bradycardic it may be best to consider thermal therapy and fluids to assist perfusion once the heart rate is elevated by the increase in body temperature. It is also worth noting that death in reptiles relates to a lack of cerebral function and this can occur prior to the cessation of the heart beat and the heart can continue to beat despite the skeletal muscle being in rigor mortis. On the other hand if euthanasia is performed the cerebral cortex can continue to receive painful stimuli for a time period (one hour or so) after the heart has been stopped by an agent such as pentobarbitone. So a heart beat does not equate to life and its absence may not equate to death.

Thus confirming death is not as easy as it may seem. A collapsed reptile or a cold reptile can have a very low heart rate which is only detectable with a dopper probe. If a reptile is clearly beyond help but you are not sure if it is dead or just in a state of extremis then, if warming fails to revive the patient, euthanasia should be performed anyway. Pentobarbitone should be used by injection to lead to cardiac and respiratory arrest. This can be intravenous or intracardiac. I prefer to check the heart has stopped with a Doppler and then pith the central nervous system. This is typically performed via the choana, but placing the needle via the foramen magnem or the nares is also commonly performed. Tell the owner you are going to do this. If you are not sure the reptile is dead place it somewhere warm overnight and assess it for rigor mortis in the morning.

Immobilisation of injuries

Keeping reptiles away from predators and immobilising and bandaging painful areas can assist in limiting pain impulses or perceived pain from injuries. Traditional methods of immobilisation and dressing materials can be applied to reptiles. The use of topical treatments such as silver sulphasalazine cream in a hydrocolloid gel can aid healing.