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Small Ruminant and Camelid Critical Care Tips and Tricks

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Abstract

Small ruminant and camelid veterinary medicine can be an important and economically viable portion of veterinary practice. The evolution of small ruminant and camelid owners has put veterinarians in the position to be expected to provide more extensive care for some of these patients. The New World Camelid industry continues to grow in the United States, although it is uncertain how long this growth will be sustained. The sale prices for New World Camelids remain high and due to the investment nature of these animals, owners will often seek high-level veterinary care. In addition, owners are obligated to seek appropriate medical treatment of animals with mortality insurance and this may necessitate more extensive medical care. There is also an increase in the number of owners raising pet livestock, in particular sheep and goats. These clients will often pursue veterinary medical care that is in excess of the monetary value of the animal. For these reasons, it is important for practitioners working with these species to be familiar with basic critical care techniques.

The methods described are developed from the collective experiences of clinicians at the Colorado State University James L. Voss Veterinary Teaching Hospital. While these methods are biased toward the types of routine and tertiary cases that we see, they will provide the general practitioner with many tools to treat routine and critical care patients. The topics and key concepts presented can be put to use immediately in a veterinary practice.

Résumé

La médecine vétérinaire appliquée aux petits ruminants et aux camélidés peut être une option importante et économiquement viable pour les praticiens vétérinaires. Les changements survenus chez les producteurs de petits ruminants et de camélidés font

en sorte que les vétérinaires sont en plus grande demande pour soigner ce type de patients. La production de camélidés sud-américains continue de s'accroître aux États-Unis bien qu'on ne sache pas si cette croissance va durer encore longtemps. Le prix de vente des camélidés sud-américains est encore élevé mais comme ce type d'animaux donne des retours à long terme, les propriétaires vont souvent demander des soins vétérinaires de haute qualité. De plus, les producteurs sont obligés de traiter adéquatement les animaux qui sont assurés pour la vie entraînant des soins médicaux plus extensifs. Il y a aussi un accroissement du nombre de propriétaires qui élèvent du bétail d'accompagnement comme les moutons et les chèvres. Ces clients vont souvent demander des soins vétérinaires qui excèdent la valeur monétaires des animaux. Par conséquent, il est important pour les praticiens qui travaillent avec ces espèces d'être familiers avec les techniques de soins critiques de base.

Les méthodes que nous décrivons ont été développées grâce à l'expertise acquise par les cliniciens de l'hôpital vétérinaire James L. Voss de la Colorado State University. Bien que ces méthodes soient biaisées en fonction des cas de routine et des cas tertiaires que nous recevons, ces méthodes vont fournir au praticien généraliste plusieurs outils pour traiter les patients de routine et les patients exigeant des soins critiques. Ces sujets et les concepts clés peuvent être mis à l'essai immédiatement en pratique vétérinaire.

Introduction

The types of critical care cases often seen by small ruminant and camelid practitioners include:

- Acute systemic diseases resulting in shock or dehydration.
- Diseases resulting in serum protein loss or decreased serum protein production.
- Acute or chronic blood loss.
- Upper airway obstruction.

- Nutritional and metabolic disease resulting in cachexia or inappropriate fat mobilization.
- Gastrointestinal diseases resulting in fluid loss or gastrointestinal dysfunction.

Key factors in the treatment of these diseases is re-establishment of normal systemic homeostasis, maintenance of hydration and fluid balance, maintenance of a functional airway, providing optimum oxygen delivery to tissues, and providing nutritional support. The following topics will provide useful guidelines for effectively addressing these problems in the critical care patient.

Intravenous Catheters

Having a direct line for intravenous access is very important in managing patients requiring repeated blood samples, parenteral treatments, and intravenous fluid support. The two most common sites used for intravenous catheters in small ruminants are the ear and jugular veins. In general, intravenous ear catheters are most easily placed and maintained in goats, followed by sheep and then llamas and alpacas. The potential benefits of an ear catheter are ease of placement if a satisfactory vein can be identified and the fact that you do not need to shave the neck. The main disadvantage is the limited size of the catheter (20-18 gauge) that can be used. Ear catheters are perhaps best used if you need limited IV access with small volumes of fluid therapy. Ear catheters work well if you want to give repeated doses of IV medications, and these can be managed well by owners with limited instruction.

Placement of Ear Catheters

It is recommended that the ear is clipped and prepared for aseptic catheter placement. Exam gloves or sterile gloves can be used for placement of the catheter. The vein can be held off by an assistant or with a rubber band secured around the base of the ear. While there are many ways to stabilize the catheter in the ear, we find that placing a tape butterfly on the catheter and then securely taping this to the ear works well. A roll of gauze inside the ear forms a nice structure for this wrap. Extension sets or fluid drip sets should be secured at another site (horn, head and neck, halter) to help prevent pulling out the catheter if the lines become caught or tangled. If you are very concerned about keeping the catheter protected, a stockinette bandage can be placed over the head and neck to keep extension sets and IV lines under control.

Placement of Jugular Catheters

The jugular vein is very superficial in goats and sheep and is easy to visualize, providing the neck is shaved. It is often thought that the jugular vein is deep in the neck of llamas and alpacas, but the primary prob-

lem is the thickness of the skin and not the depth of the jugular vein. In fact, once the catheter passes through the skin of the jugular furrow, it often requires just a short, sharp thrust to enter into the jugular vein in all of these species. Another hint on jugular catheters is that the wall of the jugular vein can be quite tough and will roll away from the stylet of the catheter if you try to ease it into the vein. Thus, it is important to line the catheter up with the jugular vein and quickly thrust it through the vein wall at a fairly sharp angle. Common catheter sizes are 18-16g x 1 ½ to 3-inch catheters for neonates and 16-14g x 3 to 5-inch catheters for adults.

Once the jugular furrow is clipped and aseptically prepared, a local block is performed over the catheterization site with 0.5 to 1ml of 1-2% lidocaine using a 3 ml syringe and 25-gauge, 5/8-inch needle. Try to minimize the amount of lidocaine used, as excessive subcutaneous lidocaine will effectively collapse the jugular vein just as a hematoma will. After the site is blocked, make a small stab incision through the skin with a #15 scalpel blade. The skin incision is very helpful and helps decrease collapse of the jugular due to compression from skin drag. The skin can either be pinched up away from the jugular or slid dorsally over the neck muscles to make this incision and avoid incising the jugular vein. In sheep and goats, a simple stab incision is satisfactory. In llamas and alpacas, particularly intact males with thick skin, it is useful to make a longer (about 1cm) incision that is angled down through the skin in the same plane and direction that the catheter will take. With the thick skin, it can be difficult to pinch the skin or move it dorsally to protect the jugular vein. Another trick used in llamas and alpacas is to take a 20-gauge needle and bend it into a slight curve. This needle is then inserted horizontally through the skin just above the catheterization site and is used to tent the skin away from the jugular vein. Make sure you "feel" the incision go all the way through the skin as any drag of the catheter in the skin will make it more difficult to insert.

In placing the catheter, hold the jugular at the proximal neck and allow time for it to distend. In severely dehydrated or hypotensive animals, this may take time. You can also try to distend the jugular by milking blood down to it. Sometimes the jugular will fill better when the animal is in lateral recumbency, and a last trick may be to allow the head of the animal to hang over the edge of an exam table while it is in lateral recumbency. Hold the catheter horizontally and flush it out with some sterile heparinized saline (1ml 1000 Units/ml heparin in 250ml isotonic saline). The catheter is held by the hub with the index finger over and occluding the end of the hub (like a dart). When inserting the catheter, first place it through the stab incision and make sure that there is no skin drag. If

there is any skin drag, then set the catheter to the side and deepen or extend your skin incision. Once the catheter is through the skin, line it up with the distended jugular at an angle of 45-60 degrees and give a quick, short, forceful insertion. Lift your finger off the hub and watch for a flash of blood. If the blood comes up to the hub, place your finger back on the catheter, flatten it out a bit and insert the stylet and catheter another 1 cm. Check for blood again and if you are still in the jugular vein, slide the catheter off the stylet down the jugular vein and finally remove the stylet. You should feel minimal resistance to passing the catheter and many times you will recognize a successful catheterization by the feel. If the animal is markedly hypotensive, you may not observe a flash of blood into the catheter hub. This is more common if the animal is standing or in sternal recumbency than if the animal is in lateral recumbency. In these cases, it may be useful to have an assistant attach an extension set and syringe filled with heparinized saline to the catheter and provide slight periodic aspiration as you are trying to perform the initial venipuncture and place the catheter.

Once the catheter is inserted, an injection port or extension set can be attached and the catheter should be checked for blood withdrawal and then flushed with heparinized saline. There are several ways of securing and protecting jugular catheters. My preferred method is to place a ½-inch butterfly on the hub of the catheter and suture this directly to the skin. This can be further secured by using a cyanoacrylate glue to attach the wings of the tape butterfly on the hub to the skin. Another important site to glue is right where the end of the hub and catheter enter into the skin. A second suture can be placed around the injection port or extension set if desired. Skin suture loops can be used to hold the extension set next to the body. Alternatively, extension sets can be held in place by small "pony tails" of hair or wool made with tape. It is very important to firmly secure the extension set to another site on the body to help prevent the catheter from being pulled out if the extension set or fluid line gets caught or tangled. When IV fluids are not running, IV catheters can be kept patent by flushing with heparinized saline every six to eight hours.

The use of neck wraps over catheters is controversial. Neck wraps can help protect the catheter in some cases, but can also cause complications if they slip and put pressure on or kink the catheter. Problems with neck wraps are more common when the wool or fiber around the neck is thick. It is my personal preference to not use neck wraps to help hold catheters in place.

Special Tricks for Llama and Alpaca Catheters

Venipuncture and jugular catheterization of llamas has been described in the literature.^{1,5} It is gen-

erally recommended that catheters are placed in the right jugular vein in llamas and alpacas. It has been suggested that catheter placement or venipuncture of the left jugular vein may be a cause of megaesophagus in llamas and alpacas. However, we have not observed any association of megaesophagus with left side venipuncture. Due to the thick skin and fiber, it can be difficult to identify jugular distension in the unclipped animal. One trick is to hold the jugular off and then release it and watch for the collapse of the jugular rather than the filling of the jugular to identify the appropriate site for venipuncture or catheterization. This also works very well in sheep. The carotid artery lies just dorsal and medial to the jugular vein in llamas and alpacas. It is not uncommon to inadvertently pass through or by the jugular vein and enter the carotid artery in these animals. When you do this, place direct pressure over the catheterization site with gauze and remove the catheter, keeping firm pressure on the site for at least three minutes. Then you can try again.

When securing the catheter to the skin, the hub of the catheter is often directed away from the skin and does not lie flat. This is due to the thickness of the skin and the angle required to insert the catheter. If an extension set is then attached and secured to the skin, it will cause the hub of the catheter to lift up and may kink the catheter where it enters the skin. A solution for this is to place a small gauze "pillow" between the extension set hub and the skin when you suture this in place. This helps keep a neutral angle of insertion and minimizes the kinking of the catheter.

Sometimes you will get an owner that is adamant that you minimize the fiber removed when clipping the neck for jugular catheter placement. A method that has worked for us is to first accurately identify the jugular furrow and site of catheter insertion by performing a venipuncture with a 23-gauge, 1-inch or 25-gauge 5/8-inch needle. The needle is then removed from the jugular and left in subcutaneously, the hair is parted and the neck is wrapped over this site with a bandage material such as VetWrap to compress the hair. Then a small square (1 inch square) is cut out of the bandage over the needle and the needle is removed. The hair is clipped or shaved and the site is prepared for catheterization as above.

The vascular valves found in the jugulars of llamas and alpacas can sometimes impede the ability to thread a catheter down the jugular vein. When this happens, you often feel that the catheter is inserting normally into the jugular but then meets resistance. The catheter can then kink back upon itself or be directed outside of the vein. The problem is more common with longer (>5 inch) catheters. There is no single trick that will help remedy this situation. Tricks that have worked with variable success include:

- Keeping the jugular held off and flushing with heparinized saline while threading the catheter down the vein.
- Using a shorter catheter. The disadvantage of this is that shorter catheters are more likely to slip out of the jugular vein.
- Only advancing the catheter just beyond the tip of the stylet and then threading both the stylet and the catheter down the jugular together.
- Moving the insertion site higher in the neck or lower at the site where the valve appears to be.
- Use an over-the-wire catheter or insert a regular catheter over a J-wire.

Intravenous Fluid Treatment

There are numerous resources on fluid therapy in livestock.^{2,3,11,12} From a clinical perspective, the decision to administer intravenous fluids and expand fluid volume is far more important than the specific choice of fluid type and rate. Clinical signs that support the administration of intravenous fluids include:

- Evidence of systemic shock such as weakness, tachycardia, cold extremities, capillary refill time >3 seconds, and pale mucous membranes.
- Evidence of dehydration such as eyeball recession, dry mucous membranes, or prolonged skin tent.
- Lack of voluntary oral fluid intake.
- Significant abnormalities in serum electrolyte concentrations.

Fluid choice can be simply broken down into three categories. The majority of patients will respond adequately to any balanced electrolyte fluid. Patients with gastrointestinal obstruction or stasis often have a hypochloremic metabolic alkalosis and will respond best to isotonic saline. Sodium bicarbonate may be used as either isotonic sodium bicarbonate (1.3%) or added to balanced electrolyte solutions in patients with a metabolic acidosis. Common causes of metabolic acidosis in small ruminants and camelids include absorption of D-lactate from the gastrointestinal tract (rumen or C1 acidosis, enterocolitis, fading kid syndrome), sodium loss due to secretory diarrhea, sepsis or other causes of systemic shock leading to L-lactate accumulation from poor tissue perfusion. The amount of bicarbonate to administer to a patient can be calculated as:

Total Body HCO₃ Deficit =
[Base Deficit (mEq/L)] x [BW (Kg)] x [0.5]
 • Base deficit can be estimated as 24-total CO₂

Useful Measurements for Bicarbonate:

- 8.3 g NaHCO₃ (baking soda) = 100 mEq HCO₃⁻
- 8.3% NaHCO₃ contains 1 mEq HCO₃⁻/ml

- 1 cc solid NaHCO₃ (baking soda) = 12 mEq HCO₃⁻

The amount of fluids and fluid rate administered is determined by the degree of hypotension due to systemic shock or hypovolemia (dehydration), the maintenance needs, and the extra losses for the patient. There is no standard method of estimating dehydration in small ruminants or camelids. While skin tent can be reliable in goats, it is often difficult to assess skin tent in sheep and camelids due to fiber length, and in camelids due to skin thickness. Similar to what has been described in calves⁴, it appears that eyeball recession is a reasonable indicator of hydration status in small ruminants and camelids. The following simple formulas give clinically useful estimates of hydration status in small ruminants and camelids.

Estimating Dehydration

- % Dehydration ≈ eyeball recession (mm) x 2
- % Dehydration ≈ (skin tent seconds x 2) - 4

Fluid volumes and rates are determined by the clinical status and hydration of the patient. Boluses of larger volumes of fluids are beneficial for animals in shock from sepsis, blood loss, or trauma. Rehydration fluids may be administered rapidly to dehydrated animals, but then the rate should be reduced to account for maintenance plus additional losses. Prolonged administration of high-volume fluids can result in over-hydration and other medical complications. Simple and effective fluid volumes and rates are listed below.

Recommended fluid volumes and rates are:

- **Shock Fluids:** 80-90ml/kg given as ¼ boluses to effect.
- **Rehydration Fluids:** Estimated % Dehydration X Kg BW given either as a rapid bolus or over a period up to 6 hours.
- **Maintenance Fluid Rate:** 3ml/kg/hr (range 2-4ml/kg/hr)
- **Extra Fluid Losses:** Up to 5-10% BW/day in animals with severe diarrhea

Fluid drip rates can be easily calculated based on the drip volume of the drip set.

- **10 drop/ml drip set** (Less Common)
 - 1 drop/sec = 360 ml/hr
 - 3 drop/sec ≅ 1 L/hr
- **15 drop/ml drip set** (Most Common)
 - 1 drop/sec = 240 ml/hr
 - 4 drop/sec ≅ 1 L/hr
- **60 drop/ml (pediatric) drip set**
 - 1 drop/sec = 60 ml/hr

Glucose supplementation is an important aspect of fluid therapy in hypoglycemic, malnourished, or anorectic animals. Blood glucose levels can easily be measured with human blood glucose monitors. If glucose is contained in the IV fluids, make sure that the extension set and catheter are thoroughly flushed with heparinized saline before taking a sample from the catheter. It is preferable to take the sample from another vein and this can be done simply by filling the hub of a 25g needle from the opposite jugular or any other peripheral vein. The recommendation for glucose supplementation is 100-200mg/kg as a single bolus for severely hypoglycemic animals or 100-200mg/kg/hour for continuous infusion.

Dextrose Bolus:

- 5% Dextrose Bolus 2ml/kg BW
- 10% Dextrose Bolus at 1ml/kg BW
- 50% Dextrose Bolus at 0.2ml /kg BW

Maintenance Dextrose Rates:

- 5% Dextrose at 2ml/kg/hr gives 100mg/kg/hr
- 5% Dextrose at 4ml/kg/hr gives 200mg/kg/hr

Many critically ill patients are potassium depleted and may benefit from additional supplementation with potassium chloride (KCl). KCl can be safely supplemented at concentrations up to 60mEq/L in IV fluids that are administered at a maintenance rate of 3ml/kg/hr. If you are giving fluids at a higher rate, you should decrease the concentration of KCl. Routine maintenance fluids are often supplemented with 20mEq/L KCl.

Economics can still play an important role in the treatment of critical care patients and commercial IV fluids may be too expensive for the treatment of some livestock patients. Inexpensive intravenous fluids can be prepared from commercial distilled water and crystalline electrolytes. Solid salts can be added directly to 1 gallon or larger plastic containers of distilled water. Regular IV sets can be attached to the plastic containers by first making a pilot hole with a 14g needle and then driving the drip set spike into the container. Common isotonic intravenous fluids can be made as follows:

Isotonic Saline (0.9%)

- Non-iodized table salt
- 9 g (7cc) per liter
- 34 g (28cc) per gallon

Isotonic Sodium Bicarbonate (1.3%)

- Baking soda
- 13 g (13cc) per liter
- 50 g (50cc) per gallon
- 8.3 g NaHCO₃ gives 100 mEq bicarbonate

Isotonic Dextrose (5%)

- 100 ml 50% Dextrose per liter
- 380 ml 50% Dextrose per gallon

Extracellular Fluid (ECF)

- Similar to acetated ringers solution
- NaCl – 6.4 g/L, 24 g/gal
- KCl – 0.4 g/L, 1.4 g/gal
- Na acetate trihydrate – 4 g/L, 15.4 g/gal
- Final concentrations
 - 140 mEq Na+
 - 5 mEq/L K+
 - 115 mEq/L Cl-

Plasma and Whole-blood Transfusion

Excellent reviews of plasma and whole-blood transfusion are published.^{6,8,13} Plasma transfusions are most commonly used in camelid medicine for treatment of failure of passive transfer in crias since commercial llama plasma is readily available. Plasma antibodies and other plasma proteins can also improve treatment success for animals with severe sepsis. Plasma can also be useful in treating animals that are hypoproteinemic due to protein catabolism from malnutrition, intestinal protein loss, or intestinal malabsorption. It is critical in these cases to attempt to mitigate the cause of the hypoproteinemia before administration of plasma, as it will be rapidly depleted if the underlying cause is not corrected. The typical initial dose of intravenous plasma is 20-40ml/kg administered at a rate of 5-20ml/kg/hr. Since commercial plasma is not readily available for sheep and goats, whole-blood transfusions can be used at a dose of 40-80ml/kg.

Whole-blood transfusions are beneficial in treating severely anemic animals or as a source of plasma proteins when separated plasma is not available. Determining whether or not an anemic animal needs a whole-blood transfusion is based on the duration of the anemia (acute vs chronic), the animal's packed cell volume (PCV), and other clinical signs. Whole-blood transfusion is recommended when the PCV falls below 15-20% in acute anemia or 10-15% in cases of chronic anemia. Clinical signs that support whole-blood transfusion are persistently elevated heart and respiratory rate, decreased appetite, weakness and lethargy. Initial blood transfusion volume of 10ml/kg (1% BW) is recommended and may raise the PCV by 3%. It is recommended to start the transfusion at a low rate of 0.1ml/kg/hr for the first five to 10 minutes and reevaluate vital signs. If there is no evidence of adverse reactions, the rate can be increased to 20ml/kg/hr. Blood-type matching is not considered necessary for animals that have not previously received a blood transfusion due to the wide diversity of red blood cell antigen groups in

ruminants and camelids. About 75% of transfused red blood cells are destroyed in 3-4 days. However, this is sufficient time for the animal to mount a regenerative response to the anemia. Red blood cell survival following repeat transfusions is very short, and may only be a few hours.

Up to 1% BW of blood can be collected from a donor animal. The most convenient and sterile way to collect whole blood for a transfusion is to use dedicated commercial whole-blood collection bags containing acid citrate dextrose (ACD) or other preservative anticoagulant. Blood collected with ACD can be stored for up to five days with refrigeration. If dedicated whole-blood collection bags are not available, blood can be collected directly into a glass or plastic container or an emptied IV fluid bag containing an anticoagulant such as sodium citrate (100ml of 3.8% solution per liter of blood) or heparin (5000 units per liter of blood).¹³ Whole blood collected with sodium citrate or heparin is not suitable for storage and must be used immediately. A simple and economical method is to dilute the heparin in saline (200 units/ml) and place this in an emptied plastic container or fluid bag for blood collection. Collect the blood directly into the container with mixing to prevent clotting. Whole blood and plasma should only be administered to the patient using a blood administration set that will filter out precipitated particles.

Field Collection of Whole Blood for Transfusion:

- Attach an IV administration set to a liter bag of saline and drain out all but about 25-50 ml.
- Add 5000 units of heparin.
- Clip and aseptically prepare the venipuncture site of the donor animal. You may consider placing a local block at the venipuncture site.
- Wrap a gauze pad at the base of the neck to hold off the jugular.
- Perform a venipuncture with a 14g or larger needle and attach to the IV administration set attached to the fluid bag.
- Collect blood by gravity flow until the bag is full or the appropriate amount is collected.
- Change the fluid IV administration set to a filtered blood administration set before giving to patient.

Tracheostomy

A tracheostomy can be a life saving procedure when upper airway obstruction is present. Common causes of upper airway obstruction seen in small ruminants and camelids include laryngeal dysfunction, retropharyngeal swelling (abscess, tumor, hematoma, or trauma), neck trauma, and envenomation (rattlesnake,

spider). The procedure for performing a tracheostomy is well described.⁷ The procedure is performed in the ventral neck at the level of the mid trachea. If time allows, the surgical site should be clipped and prepared for aseptic surgery. A local line block of lidocaine is performed. The trachea is grasped and a 3-7 cm longitudinal incision is made through the skin over the trachea. Muscles lying over the trachea are bluntly dissected until the trachea is identified. In goats and sometimes in sheep, the trachea is generally superficial enough and can be easily isolated. However, in llamas, alpacas, and cattle, the trachea is often deep in the musculature. Isolation and visualization of the trachea can be greatly improved by placing two stay sutures into the trachea to help elevate the trachea to the skin incision. A horizontal incision about 1/3 the circumference of the trachea is made between two tracheal rings. The stay sutures now also help hold the trachea open to aid placement of the tracheostomy tube. If a designated tracheostomy tube is not readily available, one can be fashioned from an endotracheal tube, a stomach tube, garden hose, or any other suitable tubing. The tracheostomy tube can be held in place with stay sutures or gauze wrap tied around the neck. The tracheostomy tube should be removed and cleaned at least once daily. Keep the tracheal stay sutures in to help aid replacement of the tracheostomy tube after it has been cleaned. Triple antibiotic ointment can be applied to the tracheostomy site and petroleum jelly can be applied to the skin below the tracheostomy site to help prevent skin scald from the tracheal secretions. When the tracheostomy tube is finally removed, the tracheostomy site is allowed to heal by second intention.

Temporary Rumen Fistula

A temporary rumen fistula is a simple, efficient, and cost-effective means to provide access to relieve rumen distension, remove or replace rumen contents, and provide enteral nutrition, fluids, or other oral treatments repeatedly to sick or debilitated animals.¹⁰ This can be performed in small ruminants or on the first gastric compartment (C1) of camelids. The goal is to provide a small, 1-4 cm diameter fistula in the left paralumbar fossa. Once the animal has recovered, the fistula can be allowed to close by second intention or closed surgically.

To perform the procedure, the left paralumbar fossa is clipped and prepared for aseptic surgery. Either a paralumbar block or an inverted L block is performed. The fistula should be placed in the mid-dorsal aspect of the paralumbar fossa. A circular incision is made in the skin. This incision should be made slightly smaller than the intended size of the fistula as the hole in the skin will stretch open when it is completed. The under-

lying muscle layers are then dissected away, leaving a muscular ring to help work as a valve. The peritoneum is incised and the rumen or C1 wall is identified. The rumen/C1 wall is grasped with Allis tissue forceps and tented out through the hole in the body wall. Three or four mattress stay sutures are made between the rumen/C1 wall and the skin (not incorporating the muscle layers) using 2/0 or 3/0 absorbable suture. The rumen/C1 wall is then sutured to the skin in a simple continuous pattern using 2/0 or 3/0 absorbable suture in two or three sections. The placing of the suture bites should be close together (about 3mm) to provide a tight seal between the rumen/C1 wall and the skin. The rumen/C1 wall is then incised to allow drainage of the rumen/C1 contents and decompression.

If the purpose is simply to relieve chronic ruminal tympany, the fistula can be left as-is. If the goal is to provide temporary access for treating the rumen compartment, then a cannula can be fashioned from an appropriate-sized syringe case. The end of the syringe case is cut off at an angle to aid insertion into the fistula. Two holes are placed on two sides of the flanged rim of the syringe case and used to attach the syringe case to the skin by stay sutures. This syringe case cannula can be capped as needed during treatment to minimize drainage. Once the animal has returned to normal health, the cannula can be removed to allow the fistula to close on its own over three to four weeks. Alternatively, the fistula can be surgically closed in a two or three layer closure.

Enteral Feeding

The two most common conditions where we utilize enteral feeding in small ruminants and camelids are nutritional metabolic disorders (hepatic lipidosis, rumen acidosis, forestomach indigestion) and critical neonates that are not nursing appropriately. Enteral feeding of functional ruminants is difficult by stomach tube due to the consistency of the feed material and repeated oral-gastric intubation. Thus, we routinely place temporary rumen/C1 fistulas for these cases. Enteral feed mixes can then be created from any available feedstuff that can be placed through the cannula. The best material for enteral feeding in the functional ruminant is a slurry of rumen fiber and fluid collected from a fistulated rumen fluid donor cow. This can be supplemented with ground alfalfa or grass hay, alfalfa pellets, or other pelleted feeds. However, be very cautious when using pelleted feeds, as their high digestibility can very easily result in rumen/C1 acidosis. For this reason, it is recommended that the pH of the patient's rumen/C1 fluid be checked daily while feeding. The rumen/C1 fluid pH should remain between 6.0 and 7.5. The goal is to provide at least 1% and up to 2% BW

of dry matter intake per day until the animal starts eating on its own.

Repeated enteral feeding of neonates can be another important treatment tool. Supplemental enteral feeding may be needed because the dam is not lactating sufficiently due to failure to produce milk, mastitis, or other disease. Alternatively, it may be needed to supplement a weak or septic neonate. Recommended milk intake for maintenance is 10% of body weight. Neonates normally consume 15-20% of body weight for growth. Initially, this should be divided into frequent feedings every two to three hours. Over the period of one to three weeks this can be decreased to four to six feedings per day. If the dam is lactating and the neonate will nurse from the dam, then supplementation with 10% BW is generally sufficient and the neonate will get the rest from the dam. Many times, the neonate will nurse from a bottle. However, if it will not nurse from a bottle, then a temporary nasal esophageal feeding tube can be placed to provide easy repeated feeding. Red rubber feeding tubes are suitable, but economical specialized nasogastric feeding tubes are also available (Argyle Indwell Feeding Tube, Tyco Healthcare, Mansfield, MA). These tubes can be sutured in place at the nose and are easily maintained and used by the owner if the patient is not hospitalized. Neonates are able to nurse normally while the tube is in place. Establishing placement of a temporary nasogastric tube early in the treatment of a weak or septic neonate can dramatically improve nutritional support and treatment success.

Parenteral Nutrition

Parenteral nutrition can be a tremendous adjunct to treatment of chronically malnourished or cachectic patients, patients with enteric disease, or patients with hepatic lipidosis or other nutritional metabolic diseases. Small ruminants and camelids with severe intestinal parasitism can be maintained on parenteral nutrition during early treatment allowing the intestinal tract time to heal and become functional again. Often, this only takes three to five days. Parenteral nutrition is particularly helpful in treating camelids with severe *Eimeria macusei* enteritis and hypoproteinemia. Chronic malnutrition can have many causes including inadequate nutrition, dental problems, chronic gastric ulcers, or other abdominal disease. At the time of presentation these animals may be too compromised to consume sufficient feed to recover. Negative energy balance associated with late-term pregnancy or early lactation can also result in metabolic nutritional disorders including pregnancy toxemia, hypertriglyceridemia, and hyperlipemia. Parenteral nutrition can provide the necessary nutritional support to help the animal through the crisis.

Parenteral nutrition is often categorized as total or partial based in part on the number of calories supplied. The distinction is not really necessary as the goal is simply to provide sufficient nutritional components intravenously to support the patient during decreased enteral intake. Parenteral nutrition formulations generally consist of glucose, amino acids and other vitamin or mineral supplements. For patients without evidence of hypertriglyceridemia or hyperlipemia, lipids will often be included in the parenteral nutrition. Gross hyperlipemia can often be observed by simply spinning down a blood sample and evaluating the plasma. Triglyceride levels should be checked prior to instituting parenteral nutrition containing lipids, particularly in camelids. Normal triglyceride levels in camelids have not been rigorously established but reference intervals of 8.3-55.8 mg/dl are reported.¹⁵

Several parenteral nutrition protocols that can be used in small ruminants and camelids are available in the literature.^{9,14,16} A summary of the common protocols used at the CSU VTH are listed below.

Parenteral Nutrition with Lipids (Protocol 1)

- Dextrose 50% 1000ml (final 22% Dextrose)
- Amino Acids 10% 1000ml (final 4.4% Amino Acids)
- Lipid Emulsion 20% 250ml (final 2.2% Lipid)
- Caloric Density: 1.1 kcal/ml
- Target rate: 2 ml/kg/hr (53 kcal/kg/day)
- Cost: \$62.50 for 2250ml
- Lipid % may be increased as needed up to 50% of total calories (1000ml).

Parenteral Nutrition with Lipids (Protocol 2)

- Dextrose 50% 1000ml (final 14.3% Dextrose)
- Amino Acids 10% 2000ml (final 5.7% Amino Acids)
- Lipid Emulsion 20% 500ml (final 2.9% Lipid)
- Caloric Density: 0.97 kcal/ml
- Target Rate: 2 ml/kg/hr (53 kcal/kg/day)
- Cost: \$137.50 for 3500ml
- Provides high caloric density with a lower dextrose concentration and less risk of hyperglycemia.

Simple Parenteral Nutrition for Neonates with Enteritis

- Dextrose 50% 1000ml (final 25% Dextrose)
- Amino Acids 10% 1000ml (final 5% Amino Acids)
- Caloric Density: 1 kcal/ml
- Target Rate: 2 ml/kg/hr (50 kcal/kg/day)
- Cost: \$37.50 for 2000ml

Parenteral Nutrition for Camelids with hypertriglyceridemia, hyperlipemia, or hepatic lipidosis)

- Dextrose 50% 1000ml (final 12.5% Dextrose)
- Amino Acids 10% 2000ml (final 5.0% Amino Acids)
- Crystalloids (LRS) 865ml
- Calcium Gluconate 23% 100ml
- Potassium Chloride 4mEq/ml 30ml
- Vitamin B-Complex 5ml
- Caloric Density: 0.63 kcal/ml
- Target Rate: 2 ml/kg/hr (30 kcal/kg/day)
- Cost: \$90.00 for 4000ml

Low Monitoring Parenteral Nutrition

- Balanced Isotonic Fluids 4000ml
- Dextrose 50% 500ml (final 4.5% Dextrose)
- Amino Acids 10% 1000ml (final 1.8% Amino Acids)
- Caloric Density: 0.23 kcal/ml
- Target Rate: 2-4 ml/kg/hr (10.9 kcal/kg/day)
- Cost: \$55.00 for 5500ml
- Will provide 20-40% maintenance PPN for 25-55 hours for a 50kg animal.
- Advantage is that the rate is not as critical (less risk of hyper or hypoglycemia) and these fluids can be used without a fluid pump.

Parenteral Nutrition Administration and Monitoring:

- A reasonable maintenance caloric goal is 50 kcal/kg/day.
- Start at ¼-1/2 target rate for 6 hours, then check blood glucose.
- If blood glucose <200 mg/dl, continue to increase rate by ¼ every 6 hours until target rate is reached.
- After acclimation, want to maintain blood glucose under 150 mg/dl at maintenance.
 - Can monitor urine glucose as an alternative since renal glucose threshold is about 180 mg/dl.
- Wean off by decreasing rate by ¼ target rate every 6 hours to minimize risks of hypoglycemia.
 - Blood glucose should remain >70mg/dl.

Components for parenteral nutrition can be purchased from many veterinary and human health distributors. Care should be taken when mixing parenteral nutrition components to ensure sterility. It is best to compound these aseptically in a biological safety hood. However, this is rarely available in a private practice setting, and a quiet spot on a clean counter out of the

way of traffic will do. While commercial parenteral nutrition bags are available, these are expensive and generally not necessary. It is generally simpler and more economical to drain crystalloid fluids from a 1, 3, or 5-liter commercial fluid bag and then refill with the parenteral nutrition components. Swab all stoppers with isopropyl alcohol and then connect an IV fluid administration set to the component to add and insert the other end into the injection port of the empty fluid bag. Deliver the appropriate volume and then repeat for the other components. The lipids should be added **LAST** after the other solutions because they otherwise may precipitate. Try to mix only what will be used in a 24-hour period. Any extra fluids should be refrigerated for up to 48 hours.

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