Anesthesia and Common Surgical Procedures in Small Ruminants
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Preanesthetic considerations
Small ruminants, as well as other ruminants, are susceptible to complications associated with anesthesia and recumbency. Positioning of these animals in dorsal or lateral recumbency for surgery allows for the weight of the abdominal viscera to shift ventrally and cranially, causing the diaphragm to be pushed further into the thoracic cavity, thereby reducing the functional residual capacity of the lungs. As a result, increased ventilation/perfusion mismatch may lead to significant hypoventilation and hypoxemia during anesthesia. In addition, the weight of the abdominal viscera may compress great vessels such as the vena cava leading to decreased venous return, cardiac output, and arterial blood pressures. Therefore, close monitoring of cardiovascular and pulmonary functions and institution of appropriate treatments to ensure normal arterial blood pressure and adequate ventilation are important parts of perioperative anesthetic management.

Regurgitation and aspiration of stomach content can occur in farm animal species during anesthesia, especially in non-fasted animals. The risk of regurgitation decreases significantly when water is withheld for 6-12 hours and feed is withheld for 12-24 hours prior to anesthesia in small ruminants. Domesticated ruminants have a large rumen that is usually full of liquid materials which does not empty completely even after 24-48 hours of fasting. Regurgitation can occur during either light (active regurgitation) or deep (passive regurgitation) anesthesia in ruminants despite preoperative fasting and withholding of water. If the airway is not protected, a large amount of ruminal materials can be aspirated into the trachea and reach the small airways. Aspiration of acidic stomach fluid causes immediate reflex airway closure and destruction of type II alveolar cells and pulmonary capillary lining cells. Consequently, pulmonary edema and hemorrhage, hypoxemia, and arterial hypotension develop due to a loss of alveolar and capillary integrity leading to reflex airway closure, bronchospasm, dyspnea, hypoxemia, and cyanosis. The greatest impact of aspirating rumen contents lies in the amount of bacterial microflora and solid food materials aspirated. Animals may die before an endotracheal tube can be placed to protect the airway in extreme cases. Preoperative withholding of feed and endotracheal intubation with an adequately inflated cuff immediately following induction of anesthesia are recommended in all anesthetized farm animals.

Ruminants normally salivate profusely during anesthesia. Total amounts of saliva secretion in conscious adult sheep have been reported to be 6-16L per 24 hours. In the past, anticholinergics like atropine were used routinely as part of the anesthetic induction regimen in an effort to prevent salivation. However, atropine only reduces the water content of the saliva, thus causing the saliva to become more viscous and increasing the potential of airway obstruction, particularly in neonates. If the trachea is left unprotected during anesthesia, large amounts of saliva may be aspirated. Therefore, tracheal intubation with appropriate inflation of the cuff immediately following induction should be instituted to protect the airway. Placing a sandbag or rolled-up towel under the neck of a small ruminant patient to elevate the throat latch so that the mouth opening is lower that the occiput allows saliva to escape, avoiding the potential for aspiration. This technique also helps to minimize the flow of passive regurgitation during deep anesthesia.

Preanesthetic preparation
When possible, small ruminants should be fasted for 12-24 hours and water withheld for 8-12 hours before induction of anesthesia. Preanesthetic fasting may not completely prevent regurgitation, but it will decrease the amount of solid matter in the rumen. Fasting also does not prevent bloating during anesthesia, but it reduces the rate of fermentation, thus reducing the amount of gas formation, the severity of bloating, and its effect on ventilation. Neonatal ruminants or those less that 3 weeks of age, can be treated as monogastrics. Fasting of young ruminants less than 4 months of age is not recommended because of the potential for hypoglycemia and prolonged recovery. Fasting may not be possible under emergency situations, and precautions should be taken to avoid aspiration of gastric fluid and ingesta. Prevention of regurgitation and aspiration of ruminal content can be achieved effectively by placing the small ruminant patient in sternal recumbency and endotracheal intubation instituted immediately following induction. Tracheal intubation is more difficult in small ruminants as compared to large ruminants and other species because their mouth do not open widely, the intermandibular space in narrow, and the laryngeal opening is distant to the thick base of the tongue. The technique used for tracheal
intubation of small ruminants is easier when the animal is placed in sternal recumbency immediately after induction of anesthesia. Intubation is best accomplished with the help of a guide tube/stylet and long-bladed laryngoscope (250-350 mm). Hyperextending the animal’s neck helps visualization of the larynx. A cuffed endotracheal tube should be used to provide an adequate seal between the tube and the tracheal mucous membrane so to prevent aspiration of saliva and regurgitated ruminal contents. The animal should be maintained in sternal recumbency until the cuff is inflated. Blind intubation, similar to that used in horses, has been used for intubation in sheep and goats; however, it may require multiple attempts in order to successfully place the endotracheal tube in the trachea.

Another technique described as “stick intubation” has been used effectively. With the animal in lateral recumbency, a small-diameter rod made of wood or stainless steel can be used as a stylet to stiffen the endotracheal tube. One hand occludes the esophagus, and the other hand manipulates the endotracheal tube into the trachea. Care and gentle maneuvering should be used to prevent initiating laryngeal spasm and to minimize trauma to the oral mucous membrane.

Castration

Castration of the normal, young male is among the most commonly performed surgical procedures in small ruminants. An emerging trend is to delay castration in some animals (meat goats and pets) who are fed very high concentrate diets due to concerns of urolithiasis. Although bloodless surgical techniques such as banding and use of the Burdizzo emasculatome are commonly used, it may be necessary to perform surgical castration. Surgical castration is performed by excision of the distal one half to one third of the scrotum with a scalpel blade or using a Newberry knife to leave two flaps of scrotal skin (cranial and caudal). The testicles are exposed and removed after making the skin incision of choice. Many clinicians may prefer to place ligatures on the spermatic cord or to crush the cord with an emasculator, to ensure appropriate hemorrhage control. Care should be taken in using an emasculator routinely used for larger species such as horses in that an emasculator that functions well in equine castration may not adequately crush the smaller cord of the small ruminant. Therefore, ligatures may be preferred which does not take additional time and no significant expense. The spermatic cord is then transected distal to the ligature.

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Dehorning

Kids older than 2-3 weeks of age, those whose previous dehorning resulted in the growth of abnormal horn tissue (scur), and adult goats are all suitable candidates for dehorning. Disbudding should be considered for kids less than 2-3 weeks of age. It is recommended that general anesthesia be used for dehorning in adults, particularly in males with large horns. However, sedation (e.g. xylazine) and a cornual nerve block may also be effective. Because of anatomical differences, the cornual nerve block in goats requires at least two injection sites per horn versus the one injection site commonly used in cattle. In goats, the cornual nerve is a branch of the zygomaticotemporal nerve that lies halfway between the lateral canthus of the eye and the base of the horn. The horn base in goats is also heavily innervated by the cornual branches of the infratrochlear nerve which exits the orbit at or in close proximity to the medial canthus. Because of the widespread branching, the nerve is best blocked using a line block midway between the medial canthus of the eye and the medial horn base. Alternatively, a ring block around the base of the horn may also be used for anesthesia and dehorning.

Owners should be warned that this procedure can be quite bloody. In addition, it may take 4-6 weeks to heal, result in secondary sinusitis, leave holes that never completely heal, or possibly result in brain abscess. The skin around the horns should be clipped and surgically prepared. The clinician makes a circular incision through the skin 2 mm outside of the horn-skin junction. The strip of skin between the two horns should be left intact in order to improve healing and shorten healing time. An obstetric wire is then placed into the incision and a helper/technician holds the head to prevent excessive motion. With the surgeon standing in front of the animal. The cut should be made in a rostral-ventral direction. Alternatively, a small Barnes dehorner may be used to cut or snip off the horn tissue; the cut should be made to avoid injuring the thin skull. Hemorrhage may be controlled by cautery, pressure, or pulling the bleeding vessels with hemostats. If the animal has a small horn base, the surrounding skin can be undermined and stretched over the opening created by horn removal. Closing the skin over the surgical site allows for quicker recovery but is rarely possible in adult males without removing some of the frontal bone. An antibiotic ointment can be applied and a gauze pad or other absorbable material cab be placed over each removal site. The pads can be held in place by tape wrapped around the head or by a piece orthopedic stockinette pulled over the animal’s head with holes cut for the eyes and ears. Animals may be given antibiotics, tetanus prophylaxis/antitoxin, or
Cesarean section

Cesarean section is the most common abdominal surgery performed in small ruminants. Although aseptic technique in a hospital setting is ideal for better results and fewer complications, the procedure can certainly be performed in the field with good results. Cesarean section can be performed with the animal in dorsal recumbency using a ventral midline approach, but is more commonly performed with the animal in right lateral recumbency through a left paralumbar fossa incision. Local anesthesia obtained with the use of a paravertebral or line block is adequate. One must remember to limit the dose of lidocaine used for local anesthesia to no more than 6 mg/kg of body weight. Diluting the 2% lidocaine from the bottle with an equal volume of saline will create a 1% solution, which achieves adequate anesthetization of the surgical site without causing toxicity. When necessary, a 1 part lidocaine: 2 parts saline creates a 0.5% solution may be used and has also offered adequate anesthetization. The body wall of small ruminants is relatively thin, so the local infiltration does not need to be administered as deeply as in cattle. Some animals may need mild sedation, but most patients are easily restrained in lateral recumbency without sedation. Regional anesthesia using a lumbosacral epidural is a viable alternative for cesarean section in small ruminants. The recommended dose is 2 mL of 2% lidocaine per 10 kg (22 lb) body weight. An 18- or 20-gauge, 3.8 cm (1.5 inch) needle is sufficient in most animals. Onset of anesthesia occurs within 5-15 minutes and generally lasts 60-120 minutes.

The left paralumbar fossa approach allows the dam to remain in lateral recumbency which leads to far fewer respiratory complications than restraint in dorsal recumbency. The rumen is also dorsal aspect up with this positioning which serves to help retain abdominal viscera within the abdominal cavity. Should the dam bloat in left lateral recumbency, the rumen will have more room to dilate before respiratory compromise develops, and it can be decompressed if needed. Although, experience suggests that this is not a frequent problem. If bloat does occur, the practitioner usually is wise to quickly complete the surgery and return the dam to sternal recumbency rather than being overly concerned with rumen distention. The muscular body wall is relatively thin in small ruminants in comparison to other large animal species. Thus, the utmost care is required in making the body wall incision to inadvertently damaging deeper structures (rumen). The gravid uterus usually can be well exteriorized in small ruminants to allow packing off of the uterus with sterile towels before the uterus is opened. If possible, both uterine horns should be exteriorized; although, this may be difficult in some cases (multiple large fetuses). The uterine incision is best made on the greater curvature of the uterus in an easily accessible area between the ovary and the cervix. This incision can be made over the hindlimbs of the fetus. However, it is frequently easier to make the incision over the head of the kid with care taken not to make a laceration on the face of the fetus. The practitioner is best served by making an incision over each respective fetus, but occasionally two fetuses in the same uterine horn can be removed through one uterine incision. However, it is difficult to maneuver a fetus from one uterine horn into the other to permit removal though one uterine incision. So, it is often best to make at least one uterine incision per uterine horn when fetuses are present in both uterine horns. Any placenta that is not firmly attached to the uterus should be removed before closure of the uterus. The uterine incision should be closed with absorbable suture in an inverting pattern (e.g. Utrecht, Cushing, Lembert) to achieve a fluid-tight seal. One-layer closure is usually sufficient; although, a second layer can be added if the integrity of the first layer is in question. The uterus should then be cleaned of any blood or debris before it is replaced into the correct position in the abdominal cavity. The muscular body wall is closed with absorbable suture in a manner chosen by the practitioner. Closure of each muscle layer separately is recommended, but in the interest of time, layers may be combined without detriment which will be discussed in further detail.

Tube cystotomy

Placement of a Foley (16 to 24 French) catheter into the bladder and exiting through the ventral abdomen allows for continual drainage of urine. By routing urine flow through the catheter, the urethra is allowed to rest in order to decrease inflammation and promote healing. A small skin incision is made lateral to the paramedian incision and inserts the catheter subcutaneously, where it enters the abdomen and then the bladder. A purse-string suture is placed in the bladder wall to position the Foley. A small stab incision is made in the middle of the purse-string and the balloon end of the Foley catheter is placed into the bladder, after which the purse-string suture is tightened. After inflating the Foley catheter with saline, the bladder is tacked to the body wall with minimal tension. A one-way valve can be made from a finger of a latex glove and placed over the end of the catheter to create a type of Heimlich valve which helps decrease the incidence of ascending infections. The celiotomy site should be closed in three layers with an absorbable suture, and the subcutaneous tissues and skin closed in routine fashion. Sutures can be removed in 10 to 14 days. Many goats will attempt to pull or chew the tubes, so belly bandages, Elizabethan collars, and close monitoring should be used to help ensure catheter placement. Clamping the catheter should begin on the fourth day after surgery to allow for normal urination. This should be done in a dry stall and with increasing duration until a full-stream urination is achieved. Normal urination should occur for 1 to 2 days before the catheter is deflated and removed. The Foley catheter should not be removed before day 7 after surgery to reduce the chances of urine leaking from the bladder. This bladder defect is allowed to heal spontaneously.
**Bladder marsupialization**

Bladder marsupialization offers excellent long-term survival and is a good technique for non-breeding animals and those whose perineal urethrostomy sites have strictured to the point of preventing urine flow. The animal is anesthetized and placed in dorsal recumbency. The skin is clipped and aseptically prepared. An 8 to 12 cm paramedian incision is made in the caudoventral abdomen parallel and 2 to 4 cm lateral to the prepuce. The apex of the bladder is exteriorized, the bladder decompressed, stay sutures placed 4 to 5 cm apart, and the cystotomy incision is made between the stay sutures. A second abdominal incision is made on the opposite side of the prepuce. The site of the second abdominal incision is chosen to minimize urine scalding of the surrounding skin. The apex of the bladder should be pulled or lifted into the second abdominal incision by the stay sutures. All four corners of the bladder should then be sutured to the abdominal wall. The seromuscular layer of the bladder should be sutured to the fascia in a circumferential fashion using a horizontal mattress pattern with absorbable suture. The bladder margins are sutured circumferentially to the skin with an absorbable suture. The original abdominal incision is closed. Urine is voided from the bladder through the marsupialized site. Therefore, the incision should be large enough to allow urine flow but not large enough to allow bladder eversion or prolapse. Systemic antibiotics should be administered prior to surgery and for as long as 14 days post-operatively (procaine penicillin G 22,000 IU/kg IM BID, ceftiofur 2.2 mg/kg IM SID to BID). Short-term complications include bladder prolapse and cystitis, and over time fibrotic stromal closure over the marsupialization site may occur.

References available upon request.
Pregnancy toxemia

Pregnancy toxemia (ketosis) affects does during late gestation. It occurs most commonly in either fat or thin animals that carry two or more fetuses. The condition develops when the doe cannot ingest enough nutrients to meet both the glucose requirements of the growing fetus and her own body’s metabolism. During early gestation, the dam’s increased appetite is enough to encourage her to compensate for the increased nutrient needs. However by late gestation, the growing fetuses are occupying more space in the dam’s abdomen which leads to the dam being physically incapable of eating enough to meet her nutritional needs unless more nutrient-dense feeds are provided. If adequate energy is not available to the gestating doe, she can metabolize body fat to meet her own nutrient requirements. When fatty acids are metabolized at high rates, ketone bodies are produced, which can be dangerous in high levels. The condition where excess ketones are present in the bloodstream, known as ketosis, results in depression and anorexia until the doe becomes too weak to stand.

Pregnancy toxemia is a common indication for the use of dextrose-containing solutions over a period of time. In sheep and goats, a variety of metabolic derangements have been documented as part of the pregnancy toxemia, including hyperketonemia, ketonuria, metabolic acidosis, hypocalcemia, hypoglycemia, and decreased liver function from hepatic lipidosis. Hypoglycemia is an inconsistent finding in cases of pregnancy toxemia. Often, the does may have normal blood glucose or be hyperglycemic.

Pregnancy toxemia can prevented by properly managing the weight of does throughout the year, and especially prior to breeding and during gestation. Does should be body condition scored at breeding, as overweight and excessively thin does are at a higher risk for ketosis. An ultrasound can also be performed during gestation to determine fetal number. Animals gestating multiples can be fed and managed differently than those with singletons. Whenever possible, does should be divided into different groups or pens so that they can be managed differently during gestation to minimize their risk of toxemia. While it is acceptable for overweight does to lose weight during the first two trimesters, they should be gaining weight by the third trimester. Feeding grains with increased energy density during the third trimester, or about six weeks prior to kidding, will help to prevent pregnancy toxemia. Providing higher quality hay is also a good idea for gestating ewes or does.

Hypoglycemia

Hypoglycemia is most commonly seen in as a cause of weakness and depression in neonates. However, infectious causes of depression and weakness should always be considered as a potential contributing factor to these clinical signs. Hypoglycemia is relatively easy to diagnose with the aid of an inexpensive, portable glucometer. Kids typically develop hypoglycemia due to weakness, shock, environmental stress, disease of kid or dam, and poor nutrition. Administration of 50 mL/kg of dextrose (or 5% of body weight) in warm milk replacer or 1 mL/kg of 50% dextrose, either intravenously or orally (diluted to 5% dextrose), should provide ample energy to correct hypoglycemia. Intravenous administration may be necessary if gut motility is absent. If the kid does not regain an appetite, then follow-up therapy may be necessary.

Hypocalcemia

Hypocalcemia can be a problem in does most commonly shortly before or after parturition. However, the disease occurs more common after parturition. Before parturition, there is an abrupt demand for calcium in the last 3-4 weeks before parturition in does with more than one fetus due to the calcification of fetal bones. With the demands of lactation, there is an abrupt demand for calcium. The body may require 1 or days to accrue the necessary enzymes capable of mobilizing bone stores of calcium. High dietary intake of calcium, phosphorus, or some cations (potassium and sodium) decreases the production of parathyroid hormones. During decreased parathyroid function, less 1,25-dihydroxycholecalciferol is produced. Lack of this hormone results in lowered absorption and mobilization of calcium from the intestines and bones. Low dietary calcium or increased amounts of dietary anions enhances the production and release of parathyroid hormones.

Early in the course of the disease, animals most commonly present with a stiff gait, tremors, and tetany with decreased rumen motility. They may also be ataxic or constipated. As the disease progresses, animals begin to have increased heart and respiratory rates, bloat, and depression. Corneal and pupillary light reflexes are normal initially but may become depressed before disappearing entirely. Diagnosis is usually made based on history and signalment suggestive of an animal at risk of development of hypocalcemia. Serum calcium concentrations less than 4-5 mL/dL in goats are diagnostic. If animals are showing clinical signs of disease, the immediate attention is required. Intravenous administration of calcium borogluconate (50-100 mL of a 23% solution) is most commonly used. Oral calcium administration of a calcium gel designed for cattle but based on goat body weight may help prevent relapse. If using an oral calcium gel, the author has found it helpful to rinse the oral cavity with water following administration of the gel to help prevent oral irritation. Subcutaneous administration of calcium may also be used but solutions containing dextrose or other
electrolytes should be avoided, if possible, as some have been associated with abscess formation. Cardiac monitoring is necessary during treatment; therapy should be slowed or stopped if arrhythmias occur. If the treatment is successful, the animal will usually stand, urinate and/or eructate within 20 minutes.

**Metabolic acidosis**

Metabolic acidosis can be seen in goats as a result of absorption of D-lactate from the gastrointestinal tract (e.g., with grain overload or enterocolitis) and sodium loss with secretory diarrhea. Sepsis or other causes of septic shock may also cause metabolic acidosis due to L-lactate accumulation as a result of poor tissue perfusion. Animals with pregnancy toxemia may also experience metabolic acidosis. Isotonic sodium bicarbonate (1.3% NaHCO₃) and hypertonic sodium bicarbonate (5% or 8.4% NaHCO₃) may be used alone or added to other solutions to directly correct metabolic acidosis.

The base deficit is calculated by subtracting the measured bicarbonate from the normal total bicarbonate (approximate normal bicarbonate is 25 mEq/mL. The amount of bicarbonate to administer is calculated as follows:

**Neonates**

\[ \text{mEq bicarbonate needed} = \text{base deficit} \times \text{body weight in kg} \times 0.6 \]

**Adults**

\[ \text{mEq bicarbonate needed} = \text{base deficit} \times \text{body weight in kg} \times 0.3 \]

The constants 0.6 and 0.3 represent the approximate proportion of extracellular fluid volume relative to total body weight, which is different for neonates compared with mature animals. From this formula, the total milliequivalents of bicarbonate needed to completely correct the acidosis can be calculated. In some cases of neonatal diarrhea or severe grain overload, it may be necessary to administer the entire amount of calculated base deficit of bicarbonate in order to correct the acidosis. However, most cases will resolve with partial correction with administration of approximately half of the deficit over 2-4 hours. This will then be followed by complete correction of the deficit after fluid therapy which allows the normal physiologic compensatory mechanisms to function in the animal.

References are available upon request.
The basics: *Haemonchus contortus* and *Trichostrongylus* sp.

Internal parasites of small ruminants are one of the most significant health problems facing both owners and veterinarians. *Haemonchus contortus* and *Trichostrongylus* sp. are the two nematode parasites that are commonly identified on routine fecal examinations. *Haemonchus contortus* pierces the mucosa of the abomasums and causes anemia and protein loss. The eggs are deposited onto pasture where they develop into first and second stage larvae within the fecal pellet. The infective third stage larvae migrate up grass blades where they become suspended in dew droplets and are then ingested by the small ruminant. These larvae then mature into adults in the abomasum. The adult parasites live in the abomasum with each adult can produce 5,000 to 10,000 eggs per day. The pre-patent period, or time from ingestion of the third stage larvae to the time mature adults produce eggs that are passed in the feces, is about 21 days. Clinical signs of infestation with *H. contortus* may include unthriftiness, rough hair coat, pale mucous membranes, submandibular edema, and death. Diarrhea is not a common clinical manifestation of infestation with *H. contortus.*

*Trichostrongylus* sp. inhabit the small intestine and cause diarrhea and protein loss but not anemia. Many veterinarians and producers use the FAMACHA parasite control program for parasite management. However, it is important to remember that the FAMACHA program will miss severe infestations of *Trichostrongylus* sp.

Antihelmintic resistance

Parasite resistance to anthelmintics has become an increasingly common and serious problem for the last several years. Few anthelmintics are approved for use in small ruminants in the United States, and due to the tremendous investment needed by pharmaceutical companies; new discovery of anthelmintics is unlikely. The three common classes of anthelmintics used in small ruminants and camelds 1) Benzimidazoles which include albendazole (Valbazen®), fenbendazole (Safe-Guard®, Panacur®), and oxfenbendazole (Synanthic®); 2) Imidathiazole/Thetrahydropyrimidine which include levamisole (Tamisol®, Levasol®), morantel tartrate (Rumatel®), and pyrantel pamoate or tartrate; and 3) Macrolides which include ivermectin (Ivomec®), moxidectin (Cydectin®), eprinomectin (Eprinex®), and doramectin (Dectomax®). Only four drugs have been approved by the Food and Drug Administration (FDA) for use in goats. These drugs include morantel, thiabendazole (no longer marketed), fenbendazole, and phenothiazine. The FDA does allow for extra-label use of other anthelmintics as an exclusive privilege of the veterinary profession and only when a *bona fide* veterinarian-client-patient relationship exists and an appropriate medical diagnosis is made.

Resistance to benzimidazole, levamisole, and other imidazothiazole anthelmintics has been documented for many years. Even more recently, documented cases of resistance to ivermectin and other macrolides has been reported with increasing frequency. Although moxidectin (Cydectin®) has been considered the drug of last resort in many cases there has been a recent increase in the number of cases with resistance to moxidectin. It is important to remember that resistance to one drug in a class usually means resistance to all drugs in that class. The efficacy of moxidectin in the face of resistance is only due to increased potency. Therefore, it is necessary that both producers and veterinarians practice prudent use anthelmintics and learn alternative practices for controlling intestinal parasites in small ruminants and camels.

Resistance to anthelmintics has become a problem for many reasons. Some reasons include inappropriate dosing and administration of anthelmintics including rotational use of anthelmintics, and inappropriate use of pasture management after anthelmintic use.

Diagnosing resistance

A quantitative method to determine parasite burden and aid in diagnosis of resistance is extremely important. The modified McMaster’s technique is one of the most commonly used tools to determine fecal egg counts and, thus, serves as a good aid in diagnosing resistance to anthelmintics. The modified McMaster’s technique is easily performed but does require a McMaster’s slide. When determining resistance, a modified McMaster’s is performed to determine the number of eggs per gram of feces. The animal is then administered a given anthelmintic, and then 10-14 days later another modified McMaster’s technique is performed. If there is no resistance to the anthelmintic administered to the animal, one should expect to see a 95-99% reduction in the number of eggs per gram of feces. If the anthelmintic was properly administered and post-treatment egg counts are still high, then resistance to the anthelmintic administered can be concluded. As previously mentioned, it is important to remember that resistance to one drug in a class usually implies resistance to all drugs in that class. It is necessary to obtain a representative population on the farm due to individual animal variations.

Another method used to diagnose resistance of anthelmintics is a larval development assay or DrenchRite® assay. This is a test that is only performed in the laboratory of Dr. Ray Kaplan at the University of Georgia. The Drench Rite® Assay is an *in vitro* test that uses collected feces to “grow” larvae from which numerous anthelmintics are tested to determine resistance. This assay is more...
expensive than the modified McMaster’s technique but provides so much more information to the practitioner and client regarding resistance issues on the farm.

Management and control of parasites

FAMACHA® is a copyrighted parasite control program that was developed in South Africa to identify severely parasitized sheep and goats. This farm-based system uses visual inspection and analysis of the mucous membrane pallor as an indication of anemia and, thus, degree of parasitism with Haemonchus contortus. In any given flock or herd, 20-30% of the animals harbor 70-80% of the parasites. The FAMACHA® program allows producers to treat only the most severely parasitized and anemic animals on a farm which reduces the number of dewormings and helps prevent the development of resistance. FAMACHA® is based on the concept of refugia. Refugia refers to all of the parasite eggs and larvae already on pasture and all the parasites in the animals that have not seen anthelmintics. Thus by only deworming the most severely parasitized and anemic animals, there is a larger population of naïve parasites that have not exposed to anthelmintics (large refugia). This is how FAMACHA® helps prevent anthelmintic resistance. The most important factor responsible for widespread development of anthelmintic resistance is the common practice of treating all animals in a herd at one time. This practice leaves no parasites in refugia which leads to resistance; meaning that the only eggs deposited onto the pasture for the next several weeks are those that survived anthelmintic treatment or the resistant parasites. Those animals needing multiple treatments should be culled from the herd. Over a period of generations, the genetics of the herd favor animals that are parasite resistant. However, it is important to remember that the FAMACHA® program should only be applied to adults because anemia in kids and lambs can progress quite rapidly. Due to the periparturient rise in parasites, decreased immunity to gastrointestinal nematodes and high nutritional demands, periparturient does and does in lactation must also be monitored very carefully.

The FAMACHA® program is easy to use but requires intensive training and education of producers. This training can be accomplished by going through a program that is given by members of the American Consortium for Small Ruminant Parasite Control (www.wormx.info). Dates for training workshops and additional information can be found at www.scsrpc.org. It is important to remember that the FAMACHA® program is only evaluates the level of anemia due to Haemonchus contortus. Therefore, periodic fecal examinations are necessary to evaluate the level of infestation with other parasites.

Grazing practices

Another extremely important point that is imperative for good parasite control involves preserving pastures. Ideal stocking rates for small ruminants is 6-8 head per acre. If enough pasture is available, producers should remove animals from the pastures for 3-6 months to allow the larvae on pasture to die. Other practices that to reduce parasite burdens include alternating or co-grazing pastures with horses or adult cattle, and after deworming, keep the animals on a dry lot before putting on a new, clean pasture. In addition one should keep in mind that goats are natural browsers. Allowing animals to utilize browse areas reduces parasite transmission because the forage is further from the ground.

Nutrition

Nutrition is an important aspect of herd health that can help promote resistance to parasites. Appropriate levels of protein in the diet may be one of the most important factors because it helps promote good immune function. It is also thought that phosphorus may help protect animals from the negative effects of parasites as parasites pull phosphorus from the host. Other minerals that are important to immune function and thus may help promote resistance to parasitism include selenium and copper. The best means to ensure proper mineral intake is to offer a free-choice, loose trace mineral salt. However for sheep, it is important to remember to use trace minerals specifically formulated for sheep to help prevent copper toxicity.

Quarantine

There are only two ways that resistant parasites are acquired – they are either home-grown or they are purchased. Thus, management practices must also include a quarantine program that will prevent animals harboring resistant parasites from entering the farm. New additions should be isolated from the rest of the herd and placed on a dry lot without any access to forage. These new additions should then be dewormed Fourteen days after deworming, a fecal egg count or fecal floatation should be performed. The animal should only be allowed in herd if it is negative. Any parasites that remain after the deworming are resistant to at least one of the anthelmintics administered and should not enter the herd. This new addition to the herd can then be turned out on “contaminated” pasture so that any remaining resistant parasites will be overwhelmed by the local population of parasites.

How to deworm: A new way of thinking

As the effectiveness of the dewormer decreases, it provides less and less benefit, and once it falls to <50%, it is no longer useful as a sole treatment. Despite previous recommendations, rotating between dewormers will not prevent resistance from worsening. Therefore, it is no longer recommended. Instead, it is now recommended to use dewormers together at the same time in combination. There are two major benefits to using drugs in combination: 1) there is an additive effect with each dewormer used, thus the efficacy of the treatment increases with each additional dewormer given and 2) by achieving a higher efficacy, there are fewer resistant
parasites that survive the treatment. Therefore, there is a greater dilution of resistant parasites by the susceptible portion of the population. The more dewormers that are used in combination, the greater the efficacy of treatment will be. However, if all the dewormers individually have poor efficacy, the combination will not achieve a high level of efficacy. Once efficacy falls to 50%, even a combination of three dewormers will still fail to reach a 90% efficacy.

Before using combination deworming, there are a few precautions which include the following: 1) In New Zealand and Australia, products are sold that contain a combination of dewormers, so only one product needs to be administered. In contrast, in the USA, no dewormers are yet sold in this formulation, so the dewormers need to be bought and administered separately. This increases the cost as compared to the products available in these other countries. Additionally, the different groups of dewormers are not chemically compatible, thus they cannot be mixed together in the same syringe. Rather, they need to be administered separately, but can be given one immediately after the other; 2) All dewormers should be administered at the full recommended dose whether administered singly or in combination; 3) When using dewormers in combination, meat and milk withdrawal times will be equal to the dewormer used with the longest withdrawal time period; 4) If using dewormers in combination, it is critical to maintain refugia; thus, one should be using a selective treatment approach based on FAMACHA® (see FAMACHA® section of the ACSRPC website for more information on this method and for further explanations of refugia). The presence of refugia is essential to realize the full benefits from combinations. In fact, if refugia are not maintained then you will not get the necessary dilution of the resistant survivors, and this will then lead to having multiple-resistant worms that can no longer be controlled with the combination treatment; 5) If the efficacy of your dewormers are >80%, it is possible you may not notice any difference in the clinical response of treatments when applied singly vs. in combination. However, the impact on the further development of resistance could be quite large; 6) Any safety precautions that exist for a single dewormer will also exist when used in a combination; however, there are no known additional risks with using more than one dewormer at the same time.

Alternative methods and future approaches
Due to the growing and serious problem of parasite resistance to anthelmintics, alternative methods for parasite control are being considered. The ultimate goal for parasite control would be a vaccine to immunize animals against specific parasites. There are no vaccines currently in development, but this is a long-term goal. Another method includes the use of biological agents to destroy the parasite larvae. The nematode-trapping fungus, Duddingtonia flagrans, is a natural inhabitant of soil and has been used in this manner. Spores from this fungus are grown on grain and fed to the animals; the fungus passes unchanged in GI tract and concentrate in the feces. The feces are then deposited on the pasture where spores develop and trap the developing larvae within the hyphal loops. Currently this nematode-trapping fungus is not available for commercial use.

Some plants have the natural ability to suppress parasites in ruminants. Sericia lespedeza has shown the ability to reduce fecal egg counts and decrease the percentage of ova in feces that develop into infective larvae. This is a tall plant that animals browse, thus keeping their heads off of the ground and away from shorter forages where larvae can migrate up the plant. This forage contains condensed tannins which are thought to be the active compound. Condensed tannins can also be found in other plants such as Lotus corniculatum and Birdsfoot trefoil. Plants that are high in tannins may also be unpalatable to some animals due to bitterness.

However, some of the plants, particularly Sericia lespedeza, are being used with some success. Copper oxide wire particles are very potent killers of H. contortus. Animals receive capsules filled with copper oxide wire particles where they remain in the stomach and release copper over time. However, one should use caution when using copper oxide wire particles in sheep due to the possibility of copper toxicity. Currently there is no product that is available specifically for small ruminants which makes dosing difficult. Therefore, one should use caution when using copper oxide wire particles.

Another alternative that has received much attention is diatomaceous earth. Diatomaceous earth is composed of glass skeletons of diatoms and is thought to work by lacerating the cuticle of the parasite. However, no reports have described any efficacy with the use of diatomaceous earth.

References available upon request.