VME 6712C

Class 24741

I. Course information

Number: VME 6712C
Course Title: Small Animal Interventional Radiology
Semester: Fall
Course credits: 1
Students: Graduate students enrolled in UF CVM graduate program in Veterinary Medical Sciences.

II. General information

Course Instructors:

Dr. J. Brad Case (Course Coordinator)
Associate Professor
Department of Small Animal Clinical Sciences
caseb@ufl.edu
352-392-2235

Dr. Kris Cooke
Clinical Associate Professor
Department of Small Animal Clinical Sciences
cookk@ufl.edu
352-392-2235

Dr. Kat Ham
Clinical Associate Professor
Department of Small Animal Clinical Sciences
hamk@ufl.edu
352-392-2235

Dr. Penny J. Regier
Assistant Professor
Department of Small Animal Clinical Sciences
pregier@ufl.edu
352-392-2235

Dr. Frederico Vilaplana Grosso
Clinical Assistant Professor
Department of Small Animal Clinical Sciences
fvilaplanagrosso@ufl.edu
352-392-2235

Dr. Del Nero
Clinical Assistant Professor
Department of Small Animal Clinical Sciences
***@ufl.edu
352-392-2235

Maximum enrollment: 15 students

III. Course description

- This course is an introduction to current state of interventional radiology in small animal surgery (i.e., dogs and cats).

Course objectives

- Students will examine the instrumentation associated with treatment and pathophysiologic consequences of relevant diseases in dogs and cats.

- Students will define and apply specific case selection criteria and perioperative considerations for a wide variety of interventional procedures in dogs and cats

- Students will examine major steps required to perform most interventional procedures in dogs and cats

- Students will examine expected outcomes (including prognosis and complications) associated with treatment of and pathophysiologic consequences of relevant diseases in dogs and cats.

IV. Format

The course is structured in five parts: **(i)** Lectures focused on specific procedures used in selected cases; **(ii)** Description of required and recommended instrumentation as well as procedural steps in a laboratory setting. One laboratory will be held in which each student will gain experience manipulating and handling various equipment used for interventional radiologic procedures; **(iii)** Two journal club sessions for discussion and critique of selected published studies; **(iv)** One case presentation. Each student will

present a journal article with a short, guided discussion focusing on the comparative relevance between human and companion animal disease; and **(v)** A short (five-minute) case presentation by each student at the end of the course. Cases can be generated from case reports published in the veterinary or human literature, or from clinical experience of the individual student. The focus should be on the relevant pathophysiology of the disease, interventional anatomy and procedure.

V. Course Materials

The recommended textbook for this course is Veterinary Image-Guided Interventions by Weisse and Berent.

VI. Evaluation

Laboratory	25%
Journal Club 1	25%
Journal Club 2	25%
Case presentation	25%
Total	100%

Grading Scale

	Percentage
A	90 or above
A-	87-89
B+	84-86
B	80-83
B-	77-79
C+	74-76
C	70-73
C-	67-69
D+	64-66
D	60-63
D-	57-59
E	56 or below

For more information on grade points and UF grading policies, see https://catalog.ufl.edu/ugrad/current/regulations/info/grades.aspx

VII. Administrative Policies: see Student Handbook @ http://www.graduateschool.ufl.edu/media/graduate-school/pdf-files/handbook.pdf

Don't forget to evaluate your course instructors, visit the UF Evaluation site at: https://evaluations.ufl.edu/evals/

Make up and attendance policy: Please contact Dr. Case directly regarding any serious illnesses, family emergencies, or prolonged absences that result in missed work. Any

absences will require written verification. As a student, it is your choice to take all exams. If you choose to not take a quiz or exam because of another activity (work, social engagement, etc), then you will earn a zero for the grade. If an exam in this course conflicts with an exam time for another course, please contact Dr. Case as early in the semester as possible to coordinate an alternate exam time.
https://catalog.ufl.edu/UGRD/academic-regulations/attendance-policies/

Disability Resource Center
http://www.dso.ufl.edu/drc/
Students in need of accommodations should directly contact the DRC to begin the approval and implementation process. The Disability Resource Center (DRC) is located on the main UF campus and is available for students who request accommodations. OSI on the CVM campus works closely with the DRC to ensure student accommodations are met as articulated by the DRC. The DRC is able to assist students in receiving accommodations such as extended time for exams, low distraction environment, test reader or scribe and note-taking services. Please contact the DRC for more information on their processes and requirements (352-392-8565).
Students are expected to provide feedback on the quality of instruction in this course by completing online evaluations at https://evaluations.ufl.edu on the final day of the clerkship. Summary results of these assessments are available to students at https://evaluations.ufl.edu/results/.

UF students are bound by The Honor Pledge, which states, "We, the members of the University of Florida community, pledge to hold ourselves and our peers to the highest standards of honor and integrity by abiding by the Honor Code. On all work submitted for credit by students at the University of Florida, the following pledge is either required or implied: 'On my honor, I have neither given nor received unauthorized aid in doing this assignment.'" The Honor Code (http://www.dso.ufl.edu/sccr/process/student-conduct-honorcode/) specifies a number of behaviors that are in violation of this code and the possible sanctions. Furthermore, you are obligated to report any condition that facilitates academic misconduct to appropriate personnel. If you have any questions or concerns, please consult with the instructor.
Contact information for the Counseling and Wellness Center:
http://www.counseling.ufl.edu/cwc/Default.aspx, 392-1575
Contact information for the University Police Department: 392-1111 or 9-1-1 for emergencies.
Your well-being is important to the University of Florida. The U Matter, We Care initiative is committed to creating a culture of care on our campus by encouraging members of our community to look out for one another and to reach out for help if a member of our community is in need. If you or a friend is in distress, please contact umatter@ufl.edu so that the U Matter, We Care Team can reach out to the student in distress. A nighttime and weekend crisis counselor is available by phone at 352-392-1575. The U Matter, We Care Team can help connect students to the many other helping resources available including, but not limited to, Victim Advocates, Housing staff, and the Counseling and Wellness Center. Please remember that asking for help is a sign of strength. In case of emergency, call 9-1-1.

VIII. Office hours
Please contact the course coordinator caseb@ufl.edu for an appointment.

IX. Course Schedule and Presenters
Location: CVM SAH Banfield Room B (third floor)
Time: 8.30am to 9.20am (Tuesday).

Date	Topic	Instructor
8/20	Introduction to the course: Instrumentation for interventional radiology	Dr. Case
8/27	Laboratory: Scopes, catheters, wires, stents and embolic materials	Dr. Case
9/3	Radiology safety and technologies: fluoroscopy, radiography, CT and ultrasound	Dr. Grosso
9/10	Tracheobronchoscopy & tracheal/bronchial stenting	Dr. Cooke
9/17	Nasopharyngeal stenosis and stenting	Dr. Ham
9/24	Endourology; obstructed ureters & ectopic ureters in dogs	Dr. Cooke
10/1	Subcutaneous ureteral bypass systems & Stents	Dr. Ham
10/8	Fluoroscopy-assisted ureteral surgery; ureterotomy & ureteroneocystostomy	Dr. Williams
10/15	Journal Club 1: Comparative (human/feline/canine) urinary interventions	Dr. Regier
10/22	Gastrointestinal interventions: feeding tubes & decompression	Dr. Regier
10/29	Arteriovenous malformations: fluoroscopy-assisted and transarterial embolization	Dr. Case
11/5	Intrahepatic portosystemic shunts; transvenous and fluoroscopy-assisted surgery	Dr. Case
11/12	Journal Club 2: Comparative hepatic interventions	Dr. Case
11/19	Common cardiovascular interventions: PDA, PS and bradyarrhythmias	Dr. Del Nero
11/26	Lymphatic interventions: chylothorax	Dr. Case
12/3	Case presentations	Dr. Case

Lecture 1

Instrumentation for Interventional Radiology (IR)

J. Brad Case, DVM, MS, DACVS
ACVS Founding Fellow, Minimally Invasive Surgery
Associate Professor, College of Veterinary Medicine
University of Florida, Gainesville, Florida, USA

OBJECTIVES

- **To know common terminology & instrumentation of IR**
- **To understand the properties and uses of common IR devices**
- **To understand the benefits and general indications for IR**

Interventional radiology is the discipline of using radiography (fluoroscopy and CT) to guide the placement of instrumentation and implants within a patient. Although fluoroscopy is the most common technology used in IR, other non-radiography-based modalities are also used and considered synonymous with IR. Examples include ultrasonography, MRI and endoscopy. The obvious advantage of using imaging to guide placement of devices in a patient is the accuracy of placement and the reduction/elimination of the need for tissue dissection to isolate the anatomic region of interest. By reducing the tissue damage associated with traditional surgery, complications, pain and hospitalization time can be markedly reduced in both clinical patients as well as in experimental animal model scenarios.

Living beings can be thought of as complex organic structures with both solid tissue and tubular-type anatomy. Both solid structures and organ systems with *plumbing* can be accessed or altered using IR. Common examples of solid tissues addressed with IR include: liver, kidney, adrenal glands, cardiac, and pulmonary among others. Common tubular-type systems include: vascular (arterial, venous, lymphatic), the urinary tract and hepatobiliary tract.

Over the past 3 decades, tremendous advancements in technology have resulted in the production of an infinite array of instrumentation and devices specifically designed to facilitate interventional procedures in both human and veterinary medicine. As time progresses, smaller instrumentation is being developed to further improve our ability to treat disease with IR. Although a complete description of the instrumentation and devices associated with IR is beyond the scope of this lecture and course, a basic understanding of the major instrumentation will serve as a foundation for the continued learning and eventual application of IR techniques in practice.

Instrumentation associated with vascular access

Once a patient has been diagnosed with a disease that is amenable to an IR approach and is adequately prepared for the procedure, the initial step is to obtain access to the region(s) of interest. Often this involves insertion of a needle into an organ, a sheath in a vascular structure or an endoscope or cannula in a natural orifice such as the urethra. Once these devices are placed, access is secured so that the procedure can be carried out.

For vascular access, the typical approach is referred to as the *Seldinger technique.* The Seldinger technique uses a sharp needle, guidewire and introducer/sheath to obtain safe access to vessels, hollow organs and cavities.[1]

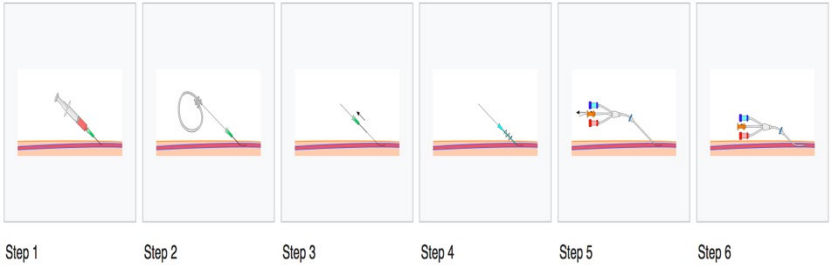
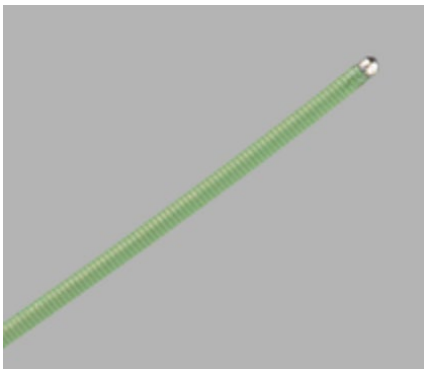


Figure 1. example of the Seldinger method for vascular access.[1]

The technique was developed (and named after) by the Swedish radiologist Dr. Sven Ivar Seldinger (1921-1998). Although the original description involved puncture of both walls of the vessel, most interventionists use a modified technique where only the near vessel wall is punctured. For example, step 1 involves insertion of a needle or catheter (typically 18 – 21G) into a vessel of interest. Next, a small guidewire (typically 0.018 or 0.035") is fed through the needle and into the vessel. Guidewires should feed smoothly and without resistance. Resistance felt during insertion indicates malposition (e.g. subintimal dissection) or deformation of the wire. Once the guide wire is advanced into the vessel, the needle or catheter is removed over the wire, leaving the wire in place. Next, a dilator/introducer sheath is fed over the guidewire and advanced into the vessel. Once the introducer is placed, the dilator is removed and the introducer secured in place with suture. Common sizes of vascular introducers used in veterinary medicine range from 4 Fr to 12 Fr. Complications associated with vascular access include malposition, hemorrhage and hematoma formation. These complications are usually avoided by careful and controlled technique. However, in some cases, hematoma formation can disrupt the ability of the interventionist to gain initial vascular access. In such instances, use of ultrasonography is necessary and is especially important if a vascular cut down is not performed or to access deeper vessels. A linear, high-frequency (e.g. 2.5 MHz) ultrasound probe is used to image the vessel of interest. Because arteries and veins typically run together in bundles, identification may be a challenge. A general rule is that the artery is deeper, smaller and non-compressible with pressure from the ultrasound probe.

Guidewires

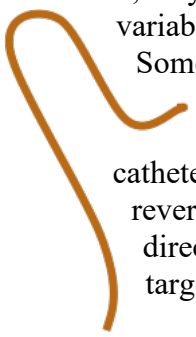
Guidewires, as their name implies, are wires that are used to guide placement of catheters and other devices into position as well as facilitate navigation of catheters into different vessels, a process referred to as *selectivity*. Guide wires typically used in veterinary medicine range in diameter from 0.018 – 0.038" with 0.035" being most common. They come in a variety of lengths ranging from 125 – 145cm. Longer (260 cm) *exchange length guidewires* are used for positioning devices such as vascular stents. The tips are either straight or angled with the angled variety being useful to navigate and select different target vessels. In addition to core diameter and tip morphology, guide wires have a variety of surface characteristics with hydrophilic and nonhydrophilic being most common. Common nonhydrophilic guidewires are composed of a steel mandrel with wire wound around the circumference and coated with polytetrafluoroethylene (PTFE). PTFE is used to reduced friction and thrombogenicity of the wire. There is a gradual taper at the



tip of the wire which makes it less thick and easier to guide into different smaller vessels. Of course there is a trade-off between stiffness of the wire and wire *pushability* which has to be considered when selecting the best wire for the application. In general, as the wire becomes stiffer, it is easier to push but harder to steer and vice versa. Two major types of non-hydrophilic guide wires are used in veterinary medicine, the Bentson and Rosen wires. **Figure 2.** Image of a 0.035" straight-tip Bentson guidewire (Cook Medical Inc). The Bentson wire has a longer taper than the Rosen wire which makes it more *steerable* and typically more desirable. Hydrophilic *glidewires* are another type of guidewire but are developed with a polymer coating that gives them a hydrophilic surface. This surface, when wet, removes virtually all the existing friction between the wire and associated catheter or vessel that the wire resides in. This makes them ideal for selecting small vessels and reducing the risk of thrombogenesis.

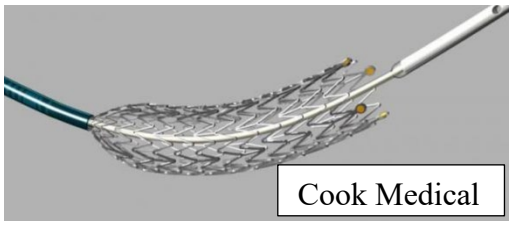
Catheters

Catheters used for IR in veterinary medicine are typically used for *Angiography, Balloon dilation/expansion or Drainage.* Depending on the application, the characteristics of the catheter vary. For example, *Angiographic* catheters are used for contrast injection as well as for embolization. They come in a variety of lengths, diameters and shapes. In general, however, they are considered straight or curved, with curved-type catheters being highly variable. For example, the number of and orientation of the curves is important. Some catheters are curved 180 degrees "Reverse curve" **(Figure 3)** and are used to select vessels at right angles often as a support catheter for additional smaller "Microcatheters" which are fed within the support catheter. Some catheters have 2 or 3 curves and are referred to as double or triple curve (or reverse-curve) catheters. These catheters often have tapered curves in opposite directions which helps "bank" the catheter off of a vessel wall and then into the target vessel.



Stents

Stents, as the name implies are cylindrical structures which act to support and/or open a vessel or luminal structure (e.g. trachea, urethra, ureter, etc). Modern stents are typically made of biocompatible metals such as nickel and titanium although some stents are synthetic (e.g. polyurethane or silicone) or biodegradable and made of different bio-inert substances. Stents can also be *covered* or *uncovered* depending on the application. Uncovered stents are used commonly in veterinary medicine for stenting of vessels (e.g. Intrahepatic Portosystemic Shunts; IHPSS) and hollow organs (e.g. urethra, trachea). In contrast, covered stents (stent grafts) are used in humans for procedures where a new vessel or connection between two vessels is needed (e.g. Transvenous Intrahepatic Portosystemic Shunt). Covered stents are also used in veterinary medicine for applications where stent ingrowth is a concern such as nasopharyngeal stenosis or post-hepatic biliary obstruction.



A further property of stents relates to the mechanism by which they are expanded. Thus, stents can further be classified as either *self-expanding* or *balloon expandable.* Self-expanding stents, as the name suggests, are stents that expand spontaneously after removal of a constraining sheath (**Figure 4)**. These stents are typically laser cut or mesh-type depending on how they are fabricated. The most significant difference between laser cut and mesh-style stents is the shortening that occurs in mesh-type stents once they are expanded. In veterinary medicine, most tracheal stents are mesh-type and shorten significantly once they are deployed. Thus, the diameter and length of the trachea need to be carefully assessed when selecting the correct stent diameter and length. If a tracheal stent is under expanded, it will be longer than if it is completely expanded. In contrast, Caval stents used commonly for stenting in IHPSS are laser cut and do not shorten significantly once they are deployed, making planning more-straight forward.

Balloon-expandable stents come as collapsed stents with rectangular shaped cells; and are fit snuggly to a balloon at the tip of a delivery catheter. When the balloon is inflated, the stent is expanded circumferentially into the wall of the target vessel. As the stent expands, the rectangular cells take on a more diamond shape. The balloon is then deflated and the delivery catheter withdrawn from the target vessel leaving the stent snuggly positioned against the wall of the vessel. Balloon expandable stents are typically used in human medicine for arterial obstructions (e.g. coronary arterial disease) and come in a wide variety of diameters and lengths. Currently, efforts are underway to develop biocompatible and biodegradable stents that dilate the artery of interest but do not maintain a permanent presence. Chronic arterial stents result in neointimal hyperplasia and restenosis which can have lethal consequences. Further, drug-eluting stents are also being developed that slowly deliver antifibrotic drugs (e.g. tacrolimus-type) to further reduce the process of or prevent neointimal hyperplasia.

Lastly, polyurethrane/silicone stents used commonly in veterinary medicine include double- and single-pigtail stents. These stents function predominantly via drainage/bypass and are thus, referred to as *drainage catheters.* The stents have coiled ends that possesses a number of inward-facing fenestrations. These fenestrations facilitate fluid/gas flow through the stent and with less risk of obstruction due to their internal position. They are deployed over hydrophilic guidewires and remain in place due to the coiled ends and proper sizing although migration and dislodgement are still possible. Common applications include ureteral stenting, nephrostomy drainage and gastrostomy drainage.

Coils

Coils are small metallic (e.g. alloy, platinum) devices ranging in size from 0.010” – 0.038” that are used to embolize vessels. They are most commonly cylindrical in morphology once deployed but other shapes (e.g. complex, tornado) are also available. Coils in the range 0.010” – 0.018” are considered microcoils and are used for applications such as thoracic duct or arteriovenous malformation embolization. Recently, microcoils have been used for embolization of IHPSS in cats and toy-breed dogs. Larger coils

(typically 0.035”) are used commonly for embolization of IHPSS in most dogs. Embolization coils come constrained within a linear delivery sheath which is inserted in to an angiographic catheter for delivery to the target. Once the coil delivery sheath is connected to the angiographic catheter, a guidewire is used to push the coil into the target. As the coil exits the end of the catheter, it takes its natural shape in place of the linear orientation it maintains within the delivery sheath. In general, coils need to be slightly larger (10-20%) than the target vessel to prevent migration unless they are supported by a stent (e.g. Percutaneous Transvenous Coil Embolization).

In summary, a vast array of devices and instruments are used in modern IR and as technology progresses this diversity is sure to expand and facilitate new techniques and advances in our ability to treat companion animal patients. Adoption and understanding of the basic instrumentation and principles of IR is the first step in developing clinical skills in the discipline.

REFERENCES

1. Seldinger SI (1953). Catheter replacement of the needle in percutaneous arteriography; a new technique. Acta Radiologica 39(5):368-76.

Lecture 2
Tracheal Collapse, Bronchial Collapse, and Stenting

Alex Gallagher, DVM, MS, DACVIM
Clinical Assistant Professor, Small Animal Medicine

Introduction

Tracheal collapse (TC) is a progressive disease of the trachea, and often lower airways, commonly seen in small and toy-breed dogs. This condition is also occasionally seen in cats. Tracheal collapse usually starts with weakening or laxity of the dorsal trachealis muscle resulting in displacement of the dorsal tracheal membrane into the lumen of the trachea. This is typically followed by progressive collapse of the cartilaginous rings due to chondromalacia that can result in complete obstruction of the airway. In many cases, there is concurrent collapse of the main stem bronchi (left more than right) and occasionally lobar bronchi and lower airways. Tracheal collapse is a dynamic condition whereby the degree of collapse changes during the respiratory cycle.

More recently, a condition called tracheal malformation (TM) has been described. This results in a "W" shape to the cartilaginous rings of the trachea. Unlike TC, TM is a static or fixed change in the rings which does not change during the respiratory cycle. This condition is most often seen in young to middle aged Yorkshire terriers.

Clinical findings

The classic clinical sign of TC is a "goose honking" cough. The cough is most notable during times of excitement or exercise. Cough induces collapse and inflammation which leads to further coughing in a vicious cycle. Stertor or inspiratory stridor may be present based on the degree and location of the collapse. As TC progresses, dogs may develop episodes of dyspnea. In some case of TC and most cases

of TM, respiratory distress may be the only clinical sign. Some cases may have other current causes of dyspnea or cough including brachycephalic airway syndrome, laryngeal collapse, laryngeal paralysis, epiglottic retroversion, or chronic bronchitis. Given the breeds predisposed to TC, compression of the left main stem bronchus due to left atrium enlargement resulting from degenerative mitral valve disease is also common and may contribute to clinical signs.

Diagnosis

Diagnosis of TC starts with a clinical suspicion based on breed, age, and clinical signs. Thoracic and cervical radiographs may show evidence of TC. Radiographs should be taken on both inspiration and expiration to evaluate for collapse along different segments of the trachea. Collapse is most common at the thoracic inlet but may involve any segment or the complete trachea. Evidence of bronchial collapse may be seen. In a small proportion of cases, tracheal collapse may appear as a soft tissue mass within the trachea. This is due to axial rotation of the trachea resulting in a widened appearance of the tracheal walls with increased soft tissue opacity between them when viewed on a lateral projection. This is most often seen in the region of the thoracic inlet. Radiographs can help assess for other causes of respiratory signs including bronchial disease, pneumonia, left heart enlargement, or congestive heart failure. Radiographically, TM will appear as a dorsal deviation of the ventral tracheal wall on a lateral projection. This typically occurs at the thoracic inlet region. There may also be concurrent TC.

Video fluoroscopy allows for real time evaluation of the trachea during both normal respiration and after induction of a cough. It allows the clinician to determine the

extent of tracheal collapse and can help assess for bronchial collapse. Recent studies have shown that up to a 50% reduction in bronchi lumen can occur in normal dogs during cough. Hence, care should be taken to not over interpret findings on fluoroscopy. A fixed or static narrowing of the trachea may indicate TM but may be difficult to distinguish from tracheal stenosis, especially if it is a short length lesion.

Tracheobronchoscopy allows the best visualization of the severity of tracheal collapse. It also allows for evaluation of the lower airways and collection of samples for cytology and culture. Up to 85% of TC dogs have a positive culture at the time of stent placement, though this may represent normal commensal organisms in some cases which do not require treatment. Anesthesia is required for this procedure. At induction and prior to intubation, a functional laryngeal exam should be performed. In appropriate breeds, evaluation for components of brachycephalic airway syndrome and assessment for epiglottic retroversion should be performed.

Treatment

The choice of therapy is dependent on type (i.e., coughing vs dyspnea) and severity of clinical signs, TC vs TM, and response to prior therapy. The degree or grade of collapse seen radiographically is not a good indicator of which therapy, medical vs interventional, is warranted. Dogs whose predominate clinical signs are coughing or cough that leads to dyspnea are typically best managed with medical therapy. Dogs that have failed proper medical management or whose predominant clinical sign is dyspnea (not brought on by cough) are typically managed with interventional (or surgical)

therapy, though in some cases medical management can still be considered. In dogs with TM, interventional therapy is the main therapy due to the static nature of the narrowing.

Medical therapy

As previously mentioned, dogs with TC typically present with cough. The cough leads to inflammation and collapse in the trachea which then leads to further coughing. The goal of medical therapy is to break this cycle. Thus, the main treatments are anti-tussives and steroids. There are a number of anti-tussives available. The author prefers diphenoxylate (Lomotil, 0.2-0.5 mg/kg PO q6-24 hrs) as this opioid does not readily cross the blood-brain barrier; therefore, there is less sedation and the dose can be titrated higher. In severe cases, the dose is started near the maximum and given every 6-8 hrs until the cough is controlled and then tapered (usually after first tapering the steroids). Steroids (typically prednisone) are started at anti-inflammatory doses. In dogs with severe cough, a dose of 1.0 mg/kg/day is started initially for 2 weeks. If improvement is noted, the dose is slowly tapered to the least effective dose.

Many dogs with TC are obese. Hence, a main stay of management is weight loss using both dietary control and exercise. This can be difficult in these dogs as exercise may exacerbate clinical signs and steroids often result in weight gain. Owners should be instructed to only exercise dogs during cooler, less humid times of day. In addition, neck collars should be replaced with harnesses for walks. Environmental allergens can exacerbate airway disease and result in coughing. Allergens can include mold (air ducts, carpeting), dust, scented candles/plug-ins/detergent, etc. These should be eliminated from the household if possible.

Interventional/Surgical therapy

Non-medical therapy should be considered when medical therapy has failed or when dyspnea is the main presenting sign. In the latter, there is a mechanical obstruction of the airway due to either TC or TM that cannot be resolved with medical therapy as the physical obstruction will still remain. Originally, the main option for these cases was surgical placement of extratracheal prosthetic rings. This has largely been supplanted by the interventional placement of intratracheal stents. However, rings are still beneficial in some cases such as when collapse occurs in young dogs and the long-term risks of stent placement are not acceptable. Recently, the use of a custom made extratracheal prosthetic device in Japan has been described with good results.

Intratracheal stenting is a minimally invasive method to reestablish luminal patency in dogs with TC and TM. This method was first reported in 2002. Overtime, the method and stent materials have been refined to improve both short and long-term outcomes. Case selection is important. Tracheal stenting has been reported to worsen cough in ~50% of dogs; hence, it should not be used for coughing alone unless medical therapy has failed. Short term complications with stenting include stent migration, infection, and granuloma formation. Long-term complications include granulomas, chronic infections, stent fracture, and progressive TC.

Some of the complications can be reduced by proper stent sizing and placement. Measurements of the trachea are performed under anesthesia with a cuffed ET tube in place just caudal to the cricoid. Positive pressure ventilation is performed with a breath hold at 20 cmH_2O. The author prefers to perform these under fluoroscopic guidance

where the trachea can be monitored to ensure full expansion. A marker catheter is placed in the esophagus in order to calibrate the images prior to measurement. This is a must in order to correctly size the stent. A stent diameter that is 10-20% (or 2-3 mm) larger than the largest tracheal measurement where the stent will be positioned should be chosen.

Placement of a stent that is undersized can result in poor apposition of the stent and tracheal walls. This can result in stent migration and/or in the development of "gutters" which can be a nidus for chronic infections and subsequent granuloma formation. In some cases, gutters will form due to poor deformity of the cartilage rings even with a proper sized stent. In these cases, dilation of the stent and trachea using the cuff of an ET tube may allow for full expansion of the trachea and the stent, resulting in resolution of the gutters. Placement of a too oversized stent can result in the stent not reaching a large enough diameter to provide adequate support of the trachea. This may lead to stent fracture as well.

Traditionally, the length of trachea to stent was chosen based on the area where collapse was noted. This was in part due to early reporting of more complications with longer stents. In many dogs, there is at least some degree of collapse beyond the area noted. In addition, stent materials have improved that reduce the fatiguability of the stent and likelihood of fracture. For these reasons, the author typically chooses to stent the majority or all of the length of the trachea, from ~1 cm caudal to the cricoid to 1 cm cranial to the carina.

Current tracheal stents are typically woven from a single nitinol wire and then compressed onto a delivery device. This results in the stent being longer when it is compressed and shorter as it is deployed, known as foreshortening. One veterinary

manufacturer of tracheal stents (Infiniti Medical) provides a foreshortening chart. This allows the clinician to determine the length the stent will be based on the diameter the stent achieves. For example, a 14 mm x 85 mm stent that is deployed and reaches a full diameter of 14 mm will be 85 mm in length. If this same stent is deployed and only reaches a diameter of 12 mm, it will be 110 mm in length. Foreshortening must be taken into account when choosing a stent size.

In some cases, there may be a large difference in the diameter of the intrathoracic and extra-thoracic trachea. Placing a stent of a much larger diameter into a much smaller area of trachea can result in stent fracture. Infiniti Medical offers a Vet Stent Duality which is tapered (larger cranially and smaller caudally) for these circumstances.

For dogs with TM, it may be difficult to get expansion of that segment of trachea during positive pressure ventilation. Stent size is chosen based on measurements cranial and caudal to the region. The author typically will slightly oversize the diameter of the stent more than normal (i.e., 20-25% larger) as the tracheal lumen often is larger once the narrowing at the malformation is resolved. These cases often require balloon dilation for the stent to open the malformation. In some instances, a second stent may be required to provide enough radial force to maintain patency at the site of malformation.

Post-stent treatment

After placement, treatment is aimed at minimizing cough, reducing inflammation, and treating any infections present. Anti-tussives and steroids, as described above, are used at the higher doses for the first 30 days. If the dog is doing well, the medications can be tapered slowly to the least effective doses. Antibiotic therapy should be

administered based on culture results. Empirical antibiotic therapy may be started pending these results.

Bronchial stenting

Main stem bronchi collapse is frequently seen in conjunction with TC. As previously noted, it may also be seen in conjunction with left atrial enlargement. When bronchi collapse is present along with TC, tracheal stenting is typically performed first to see if clinical signs improve. If not, bronchial stenting may be considered. When there is collapse of airways distal to the main stem bronchi, bronchial stenting is not typically recommended as it is unlikely to relieve clinical signs, but may be considered in some cases.

The ideal candidates for bronchial stenting are dogs where main stem bronchi collapse is the only finding and is resulting in clinical signs. These dogs are typically those with mitral valve disease resulting in left atrial enlargement and subsequent compression of the left main stem bronchus. Bronchial collapse should be evaluated endoscopically to determine the location and length of collapse. Most often, the left main stem bronchus and left caudal lobar bronchus are most significantly affected.

Stent sizing is accomplished by using the endoscope to determine the length of the collapse and using a balloon catheter to assess the diameter. Anecdotally, it is reported that most dogs will require a 8-10 mm x 20 mm stent as most commonly this condition affects small breed dogs. Veterinary bronchial stents are available through Infiniti Medical. A guidewire is placed across the appropriate bronchus and the stent

placed under endoscopic and fluoroscopic guidance. Similar to tracheal stenting, anti-tussives, steroids, and antibiotics (if indicated) are used post placement and tapered.

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Lecture 3
Nasopharyngeal Stenosis and Stenting

Alex Gallagher, DVM, MS, DACVIM
Clinical Assistant Professor, Small Animal Medicine

Introduction

Nasopharyngeal (NP) stenosis is an uncommon condition that affects both dogs and cats. In dogs, stenosis is most often the result of reflux of gastric acid into the nasopharynx. This reflux typically occurs associated with either anesthesia or vomiting. In cats, the cause of stenosis is often less clear. Similar factors as in dogs likely explain some of the cases. However, other cases have no history of recent anesthesia or of vomiting. Some of these cats may have a history of upper respiratory infections, but it is unclear if these infections are the cause of nasopharyngeal stenosis.

Treatment of NP stenosis may include several different procedures. Traditionally, stenosis has been treated either with surgical excision of the stenotic tissue, dilation of the stenosis using instruments such as hemostats, or balloon dilation. Success of these treatments has been variable and related to species. Based on the literature and clinical experience, cats seem to be more amenable to a favorable outcome with these techniques. In dogs, it is a common to have recurrent stenosis when these procedures are done. This is led to the development of NP stenting techniques. For cats, stenting is often done as a temporary procedure with later removal of the stent. In dogs, we generally expect the stent to remain in place indefinitely.

Clinical findings

Clinical signs of NP stenosis are predominately due to obstruction of the nasopharynx and resulting secondary nasal infections. Animals typically present with stertor which may wax and wane related to further obstruction from mucus. Open mouth breathing may be noted, particularly when complete obstruction of the NP occurs (termed imperforate nasopharynx). When NP stenosis is related to an anesthetic event, clinical signs typically start within days to a week.

Physical exam may reveal decreased to absent airflow from the nostrils based on a cold slide test or wisps of cotton. Nasal discharge is a common finding and may range from serous to mucopurulent. Secondary bacterial rhinitis is common due to decreased clearance of secretions from the nasal cavity allowing overgrowth of commensal organisms. This may lead to mild mandibular lymphadenopathy.

Diagnosis

Imaging of the nasopharynx is required for definitive diagnosis. CT scan is the most commonly used modality and allows for assessment of the dimensions of the NP, location of the stenosis, and length of the stenosis for determining stent size. Post-contrast CT imaging can help discriminate fluid (mucus) from tissue stenosis. Post-acquisition reformatting of images into sagittal plane images is important for best assessment for stenting. Endoscopic retrograde pharyngoscopy is beneficial to confirm the presence of the stenosis and determine if it is imperforate. This is usually performed at the time of intervention. Other imaging modalities can include MRI or contrast rhinogram performed under fluoroscopy.

Treatment

The main goal of therapy is to open the area of stenosis. Over the years, many techniques have been used. In cats, surgical resection of the stenotic tissue or palatoplasty for caudal soft palate stenosis has been described with success. More commonly, balloon dilation of the stenosis is performed as a minimally invasive technique. This is performed under fluoroscopic guidance. A guidewire is passed through one of the nares, across the region of stenosis, and down the esophagus. Conversely, a wire can be passed retrograde through a retroflexed scope in the NP to exit one of the nares. Over the wire, a balloon dilation catheter is passed and centered over the stenosis. High pressure balloons (15-30 atm pressure) are recommended as low-pressure balloons (e.g., esophageal dilation balloons) may not have enough force to open the stenosis. Balloons are expanded using contrast medium under fluoroscopy in order to assess for complete dilation. Size of balloon is chosen based on prior measurements of the normal regions of the NP on CT scan. Any "waisting" of the balloon should completely resolve to indicate complete breakdown of the stenosis.

In animals with imperforate NP, a new opening must be created across the stenosis. This requires a guidewire, introducer sheath, and a long access trocar needle (e.g., renal access needle). Initially, a guidewire is passed through a nares to the area of imperforate stenosis under fluoroscopic guidance. A 5 fr or larger introducer sheath with dilator is passed to the stenosis and the dilator removed. The access needle is passed through the introducer (using it as a needle guard) to the imperforate membrane. Under fluoroscopic and endoscopic guidance (retroflex pharyngoscopy), the access needle is advanced across the membrane into the caudal NP. This is easier when there is only a thin membrane and more difficult when there is a thick area of tissue. The needle is removed and a guidewire passed as above. Typically, the membrane needs to be enlarged with a rigid dilator over the wire prior to balloon dilation as above.

Once the stenosis is open, a decision on whether to place a stent needs to be made. In cats, the stenosis may resolve with a single or multiple balloon dilations. Recently, temporary stenting using a short segment of a silicone chest tube has been described in cats. The stent is left in place for 2-4 weeks and then removed. Successful resolution of

clinical signs occurred in 14 out of 15 cats. At UF, we have had some success with this technique but have had some cats develop a new stenosis rostral to the stent.

In dogs, NP stenosis recurs in most cases where a stent is not placed. There are reports of dogs responding to balloon dilations alone, but this is uncommon. Temporary stenting similar to cats has been tried, but stenosis has recurred after removal of the stent. Hence, more "permanent" stenting is considered for dogs. Initially, this was performed with uncovered, balloon expandable metallic stents (BEMS). However, these stents often would collapse when pressure was applied to the soft palate resulting in closure of the NP. Treatment was then transitioned to self-expanding metallic stents (SEMS). SEMS come in a few varieties including uncovered (or bare), covered (usually silicone or PTFE), and removable (Allium stent).

In some cases of stenosis, fibrous tissue may grow through the stent. Anecdotally, this seems to occur most often with imperforate NP. Using a covered stent helps prevent this issue. Covered stents can be partially or completely covered. Partially covered stents usually have 5 mm of exposed stent on each end. This can be beneficial to allow tissue in growth at the ends, helping prevent stent migration, but still allow the stent to be removed in the future if needed. A fully covered, removable stent (Allium, Infiniti Medical) is currently available. Because the stent is fully covered, stent migration is a concern. A T-bar fastener system is available to help secure the stent through the soft palate.

Proper measurement for stent size is critical. Measurements are determined from CT scan. Axial images should be reconstructed into sagittal planes for best measurement of length of the stenosis. The stent length should allow 5-10 mm of stent to be cranial and caudal to the area of stenosis. If a partially covered stent is used, the covered portion needs to meet these goals. The diameter of the stent can be more difficult to determine. The nasopharynx is typically oval in shape while the stent is circular. Some clinicians will choose stent size based on the maximal height of the NP either cranial or caudal to the stenosis. The author prefers to use a height and width measurement entered into a formula to convert oval areas to circular areas and subsequently a diameter (formulas to convert oval air duct to round air duct can be found online). The diameter needs minimal oversizing in most cases as there is not a need to support keeping a structure open, such as in the trachea. Oversizing a stent can result in the development of an oronasal fistula due to pressure necrosis.

Post dilation and/or stent placement, anti-inflammatory steroids (e.g., prednisone 1 mg/kg/day) are prescribed for 2-4 weeks. If secondary infections are suspected or documented, appropriate antibiotic therapy should be administered. Chronic infections are a common long-term complication of NP stenting. Anecdotally, this may be more common when covered stents are used as the coating provides a place for biofilm adherence. Treatment of chronic infections can be difficult. Ultimately, removal of the stent may be needed, which is an advantage of using a covered stent. Stenosis may recur after stent removal requiring restenting.

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Lecture 4

Lecture 5
Endourology: Obstructed ureters and ectopic ureters in dogs

Learning objectives

- Know / understand clinical and imaging findings associated with ureteral obstruction in dogs
- Know / understand clinical and imaging findings associated with ectopic ureters in dogs
- Understand indications for minimally invasive intervention for ureteral obstruction and ectopic ureters
- Know the basic interventional approach to treatment of ectopic ureters and ureteral obstruction in dogs
- Know the risks / complications and prognosis for interventional treatment of ureteral obstruction and ectopic ureters in dogs

Supplemental reading

- Veterinary Image-Guided Interventions: Weisse C and Berent A eds. 2015 Wiley Blackewell.
    - Ch. 29: Interventional management of canine and feline benign ureteral obstructions
    - Ch. 30: Interventional management of obstructive pyonephrosis
    - Ch. 31: Interventional management of canine malignant ureteral obstructions
    - Ch. 32: Cystoscopic-guided laser ablation of ectopic ureters
- Gallagher A. Interventional radiology and interventional endoscopy in treatment of nephroureteral disease in the dog and cat. Vet Clin North Am Sm Anim 2018; 48: 843-62.
- Tong K, et al. Rigid urethrocystoscopy via a percutaneous fluoroscopic-assisted perineal approach in male dogs: 19 cases (2005-2014). J Am Vet Med Assoc 2016; 249 (8):918-925.

Ureteral obstruction

- Causes
    - Stones
    - Strictures (acquired or congenital)
    - Ligation
    - Trauma
    - Clots / blood stones
    - Neoplasia
- Clinical signs
    - Nausea / vomiting / inappetance
    - Abdominal / back / flank pain
    - Hematuria (gross / microscopic)
    - Sometimes none (serendipitous finding)

- Pollakiuria

- Clinicopathologic abnormalities
  - Azotemia
  - Hyperphosphatemia
  - Hematuria
  - Pyuria
  - Proteinuria
- Imaging
  - Radiographs – renomegaly, mineral opacities (may be normal)
  - Ultrasound
    - Hydronephrosis – anechoic or echogenic fluid
      - Renal pelvis height > 13 mm → consistent with obstruction (dogs or cats)
    - Hydroureter – with or without focal narrowing
    - Nephroliths / ureteroliths– obstructive or non-obstructive
    - Ureteral thickening
    - Bladder (trigone) mass / thickening
  - Pyelography (radiographic / fluoroscopic)
    - Retrograde
      - Most commonly done via cystoscopy in dogs
      - Risks – trauma to urethra, trauma to ureter, incomplete study (contrast unable to reach renal pelvis)
    - Antegrade
      - Most commonly done in dogs with bladder neoplasia due to inability to visualize ureterovesicular junction (UVJ)
      - Risks → renal hemorrhage, renal pelvic laceration, renal pelvic clot formation, leakage of contrast from renal pelvis (poor study)
    - Better visualization of renal pelvis and ureters than excretory urography and decreases risk of contrast-induced renal injury

- **Ureteral stents in dogs**
  - May be placed retrograde or antegrade
  - **Retrograde**
    - **Equipment needed**
      - Rigid scopes 30º (various sizes)
        - Females
        - Males (perineal access)
      - Flexible ureteroscope (7.5-8.5 Fr)

- Hydrophilic guidewires: (0.018-0.035)
- Open-ended ureteral catheter (4-5 Fr)
- Iodinated contrast (diluted 1:1)
- Ureteral stents (various diameters / lengths)
- Stent pusher (if not provided with stent)
- +/- marker catheter

- **Procedure**
  - Pass wire through working channel of scope into UVJ to level of obstruction (or renal pelvis)
  - Pass open-ended ureteral catheter over wire into ureter (ideally ~ 1/4-1/3 length of ureter to prevent backing out)
    - Make sure ureteral catheter will fit through scope working channel (or will have to take scope off the wire)
  - Remove wire and obtain urine sample for analysis and culture
  - Perform retrograde ureteropyleogram (don't over-distend the renal pelvis)
  - Replace wire through catheter and advance into renal pelvis
    - Coil in renal pelvis
    - Be careful not to perforate kidney
  - Advance ureteral catheter to level of renal pelvis (withdraw wire and obtain urine if not done previously)
  - Withdraw ureteral catheter and measure ureteral length using 1 cm marks on catheter
    - Be sure NOT to remove the wire with the catheter
    - Can also measure from contrast study if a marker catheter has been placed per rectum
  - Select stent size
    - Length based on shaft length
    - Stent should be a few cm longer than measured length
  - Wire should still be coiled in the renal pelvis
  - Place double pig-tail stent over guide wire
    - Ideally should fit through working channel of scope
    - Make sure oriented correctly (black mark is the distal [bladder] end)
  - Feed stent over wire into ureter

        - Important to keep cystoscope at the level of the UVJ to avoid buckling of the stent in the bladder
    - Advance pusher over the wire (tapered end toward the back of the wire)
    - Use pusher to advance stent into the scope
    - Watch to make sure black mark is NOT advanced beyond the UVJ
    - Once the proximal loop of the stent is well within the renal pelvis, the wire is withdrawn to the distal part of the stent (still in scope working channel)
        - NOTE if there is no coil in the renal pelvis when the black mark is seen, the stent is too short and should be traded for a longer one.
    - Withdraw the scope into the proximal urethra as the pusher catheter is advanced, pushing the distal end of the stent into the bladder.
    - If the distal end of the stent is in the proximal urethra, the scope can be used to push it into the bladder.

- **Antegrade** (obstruction d/t bladder tumor)
    - **Equipment needed**
        - Ultrasound & fluoroscopy
        - Renal access needle
            - Can use over-the-needle catheters but they have a tendency to kink
        - +/- 4 Fr. Angled angiographic catheter (Berenstein catheter)
        - +/- 5 Fr vascular access sheath
        - Marker catheter
        - T-port
        - 3-way stopcock
        - 6-12 cc syringes
        - Iodinated contrast (diluted 1:1)
        - 2 Hydrophilic guide wires (various sizes)
        - Ureteral dilator / sheath (7-8 Fr, 45-55 cm)
        - Ureteral stents (various diameters / lengths)
            - Usually using a cancer stent
                - Non-fenestrated distal shaft
    - **Procedure**

- Patient is placed in lateral recumbency with affected kidney up
- Clip dorsal paracostal / flank area and perineum (female) / prepuce (male).
- Place marker catheter per rectum
- Stab incision in skin over kidney
- Attach T-port and 3-way stopcock to renal access needle
- Attach 1 empty syringe and 1 contrast-filled syringe to 3-way stopcock
- US-guided access to renal pelvis
- Collect urine for analysis and culture (note volume obtained)
- Inject same volume of contrast for pyeloureterogram
    - This contrast study is used to measure the ureter for selection of stent length
        - Stent should be a few cm. longer than measurement
- Feed 0.035 stiffened, angle-tipped hydrophilic guide wire down ureter and into bladder
    - May encounter resistance at UVJ – try to gently manipulate across obstruction into bladder
    - If unable to gain access to bladder, advance a 5 Fr vascular access sheath over wire, into pelvis and down the proximal ureter to prevent wire from buckling
    - Once access sheath is in place advance 4 Fr angled angiographic catheter over wire and gently manipulate across obstruction
- Once wire is in bladder, direct toward trigone and out the urethra
    - May need to do contrast study to identify urethra if tumor distorts trigone
    - If unable to pass wire out urethra, can try to retrieve it with a scope and grasper or a snare
- Now have “safety (through and through) wire
- Feed ureteral dilator / sheath over wire and across the tumor and UVJ up to the level of the ureteropelvic junction (UPJ)
    - Avoid advancing the dilator (not radiopaque) through the renal tissue

- Remove dilator leaving sheath in place at the UPJ
- Feed second wire (soft, angle-tipped, 0.035") through the sheath until it coils in the renal pelvis
- Withdraw the sheath from the ureter, leaving BOTH wires in place
- Replace the dilator in the sheath and advance over the second wire (the one that is coiled in the pelvis NOT the safety wire) until the sheath is near the UPJ
- Remove the dilator
- Place the appropriately sized stent over the second wire and advance through the sheath (sheath allows the stent to bypass the obstruction)
- Feed pusher catheter over the second wire and use to advance the stent until the proximal end is curled in the renal pelvis.
- Withdraw the sheath and wire into the urethra (the wire should still be within the stent.
- Advance the pusher and push the distal pigtail of the stent into the bladder. (Pusher for cancer stent should have radiopaque tip but verify this so you know when you are getting close to the end of the stent)
- Once the stent is in place, remove the safety wire through the urethra being careful not to displace the stent.

- **Risks/Complications**
  - Intraoperative (~3%)
    - Ureteral tear
    - Ureteral perforation
  - Post-operative (~40%)
    - Recurrent infection (13-40%)
    - Tissue proliferation at UVJ (6-40%)
    - Ureteritis (6%)
    - Stent migration (4-8%)
    - Stent encrustation (4-8%)
    - Hematuria (4%)
    - Stent fracture (2%)

~20% required stent exchange

Ectopic ureters

- Clinical signs
  - Incontinence

        - May appear continuous or intermittent
        - May be less apparent in males
            - Increased pressure in prostatic urethra
            - Longer urethra
    - Pollakiuria / stranguria / hematuria
        - Secondary infection
- Clinicopathologic abnormalities
    - Hematuria
    - Pyuria
- Imaging
    - Radiographs
        - Survey – usually normal
        - Excretory urogram / vaginocystogram
            - Diagnostic 50-70% of cases
    - Ultrasound
        - Hydronephrosis
        - Hydroureter
        - +/- renal agenesis or dysplasia
        - +/- pelvic bladder
        - Diagnostic ~60-70% of cases
    - CTEU
        - Diagnostic 65-70% of cases
    - Cystoscopy
        - Diagnostic ~75% of cases
        - Allows correction at time of diagnosis
            - Intramural EU only

~65% infection at diagnosis

Operator skill

Table 3. Sensitivity, specificity, positive predictive value (PPV), and negative predictive value (NPV) in 36 ureters (18 dogs) for imaging studies and cystoscopy as computed with surgery or postmortem examination.

Study	Sensitivity (%)	Specificity (%)	PPV (%)	NPV (%)
EU	80	100	100	44
CT	91	100	100	57
UR	100	0	100	0
Cystoscopy	100	75	97	100

EU, excretory urography; CT, computed tomography; UR, urethrography.

Sammi VF. JVIM 2004; 18: 271-281

- **Cystoscopic laser ablation of ectopic ureters (CLA-EU)**

- **Equipment needed**
  - Rigid scopes 30º (various sizes)
    - Females
    - Males (perineal access)
  - Flexible ureteroscope (7.5-8.2Fr)
  - Fluoroscopy (can be done with contrast radiography)
  - Hydrophilic guide wire (0.025-0.035")
  - Open-ended ureteral catheter (4-5 Fr)
  - Iodinated contrast (1:1 dilution)
  - Various catheters (sizes dependent on patient size)
  - Laser (Ho:YAG or diode)
- **Procedure**
  - Place scope in vestibule
    - 85-90% females will have remnant paramesonephric duct
      - Will typically laser this at the end of the procedure
  - Pass scope into urethra and bladder
  - Evaluate trigone
    - Turn so angle of scope facing downward
    - Try to locate normal UVJ (s)
  - Locate EU opening
    - May be in vestibule, vagina, urethra
  - Pass wire into ureteral opening
  - Feed open-ended ureteral catheter into ureter
  - Perform retrograde ureterogram
    - 5-10 mL diluted contrast
  - Perform urethrocystogram through scope
    - 2-5 mL/kg
  - Confirm intramural EU
  - Remove scope over wire/catheter leaving them within the EU (the wire can be withdrawn from the catheter
  - Replace scope and pass laser fiber through the working channel
  - Angle laser fiber tip toward medial wall of EU
  - Laser the EU until the orifice is within the urinary bladder **OR** until ureter appears to diverge from urethra
    - If unilateral EU can use normal UVJ as estimated location
  - Repeat retrograde ureterogram and cystogram
    - Confirm ureteral orifice location
    - Look for evidence of perforation
  - Laser ablate mesonephric remnant / vaginal septum

Perform simultaneously to evaluate ureteral opening and whether EU is intramural

- Male dogs can be done with flexible scope but this is more difficult so consider perineal access.

Lecture 6

Radiology safety and technologies: Fluoroscopy, radiography, CT and ultrasound

Federico Vilaplana Grosso, LV, DECVDI
Clinical Assistant Professor, Diagnostic Imaging
Small Animal Clinical Sciences
College of Veterinary Medicine, University of Florida

Introduction:

Interventional Radiology (IR) is the intersection of radiology and surgery, minus many of the risks, costs, recovery time, and physical trauma of traditional surgery. IR uses a wide range of imaging modalities including radiography, fluoroscopy, computed tomography (CT), magnetic resonance imaging (MRI) and ultrasound to perform minimally-invasive image-guided procedures to diagnose and treat diseases in nearly every organ system. The different imaging modalities are utilized in a variety of combinations, depending on the information required and the nature and complexity of the IR procedure which is being planned. Fluoroscopy is used for guidance, control, and monitoring operations during an IR procedure. It has the advantage compared to radiography of presenting real-time images. Although there is superimposition of anatomic structures (which doesn't occur in CT and MRI), overlying structures are eliminated by using contrast media to accentuate the specific anatomy (usually blood vessels) to be studied. A major disadvantage of fluoroscopic methods is that the x-ray dose delivered to the interventionalist, staff and patient can be high. However, fluoroscopy is the most important imaging technique used in IR procedures. For this reason knowing the radiation safety rules, radiation protection principles and radiation monitoring is of vital importance. Although radiography, fluoroscopy and CT use ionizing radiation, both methods are fast and geometrically accurate. Ultrasound suffers from image quality and tissue contrast problems, but it is fast and inexpensive. MRI is less used for IR procedures in veterinary medicine.

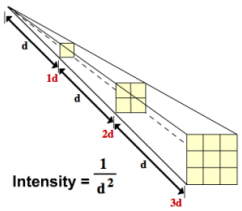
Radiation protection:

ALARA
As defined in Title 10, Section 20.1003, of the Code of Federal Regulations (10 CFR 20.1003), ALARA is an acronym for "as low as (is) reasonably achievable," which means making every reasonable effort to maintain exposures to ionizing radiation as far below the dose limits as practical.

Methods of Exposure Control
There are three principal methods by which radiation exposures to staff and patients can be minimized: Reducing **time** of exposure, increasing **distance**, and using **shielding**.

Time
Although it is obvious that reducing the time spent near a radiation source will reduce one's radiation exposure, techniques to minimize time in a radiation field are not always recognized or practiced. The imaging modalities using ionizing radiation (e.g. radiography, fluoroscopy and CT) deliver different doses of radiation depending on many factors that is necessary to consider. The time spent near a radiation source can be minimized by having a thorough understanding of the tasks to be performed and the appropriate equipment to complete them in a safe and timely manner. The amount of people during any procedure should be limited to reduce exposure. Similarly, radiation exposure to staff and patients can be reduced during fluoroscopy if the operator is proficient in the procedure to be performed.

Distance
The exposure rate from a source of radiation decreases with increasing distance from the source, even in the absence of an attenuating material. In the case of a point source of radiation, the exposure rate decreases as the distance from the source is squared, so by a factor of 4. This principle is called the **inverse square law**, where the intensity of the radiation (I) decreases in proportion to the square of the change in distance (d).



Scattered radiation from a patient, x-ray tabletop, or shield is also a source of personnel radiation exposure. For diagnostic energy x-rays, a good rule of thumb is that at 1 m from a patient at 90 degrees to the incident beam, the radiation intensity is approximately 0.1% to 0.15% (0.001 to 0.0015) of the intensity of the beam incident upon the patient for a 400 cm2 x-ray field area on the patient (typical field area for fluoroscopy). All personnel should stand as far away from the patient as practicable during x-ray imaging procedures and behind a shielded barrier or out of the room, whenever possible. The NCRP recommends that personnel should stand at least 2 m from the x-ray tube and the patient during radiography with mobile equipment (NCRP, 1989).

Shielding

Shielding is used to reduce exposures of patients, staff, and the public. The decision to utilize shielding, and its type, thickness, and location for a particular application, are functions of the imaging modality, exposure rate, and other factors. Shielding may be installed in a wall, floor, or ceiling of a room. It is incorporated behind the image receptors of fluoroscopes and radiographic machines and in the gantries of CT devices. It may be worn as protective aprons by people performing fluoroscopy. It may also be in movable barriers, such as the freestanding and ceiling-mounted shields in fluoroscopic procedure rooms. The use of a material of high density permits a thinner shield. Thus, lead is commonly used for shielding, because of its very high atomic number, density, and reasonable cost. In general, placing shielding closer to a source of radiation does not reduce the thickness needed, but does reduce the mass of shielding necessary by reducing the area of the shielding.

Aprons protect the torso of the body and the upper legs and are available in designs with only frontal shielding or with wrap-around shielding, the latter being important when the back is exposed to the scattered radiation for a considerable portion of the time. Aprons also do not protect the thyroid gland or the eyes. Accordingly, there are **thyroid shields** and **leaded glasses** that can be worn by the personnel in the room. The leaded thyroid shield wraps around the neck to provide attenuation similar to that of a lead apron. Leaded glasses attenuate the incident x-rays to a lesser extent, typically 30% to 70%, depending on the content (weight) of the lead. Unfortunately, their weight is a major drawback. Normal, everyday glasses provide only limited protection, typically much less than 20% attenuation. Whenever the hands must be near the primary beam, **protective gloves** of 0.5-mm thick lead (or greater) should be considered when use does not interfere with the dexterity required to carry out the procedure. In high workload angiographic and IR laboratories, **ceiling-mounted, table-mounted, and mobile radiation barriers** are often used. These devices are placed between the location where the x-ray beam intercepts the patient and the personnel in the room. **Mechanical supporting** or **restraining devices** must be available and used whenever possible. In no instance should the holder's body be in the useful beam, and it should be as far away from the primary beam as possible.

Lead equivalent

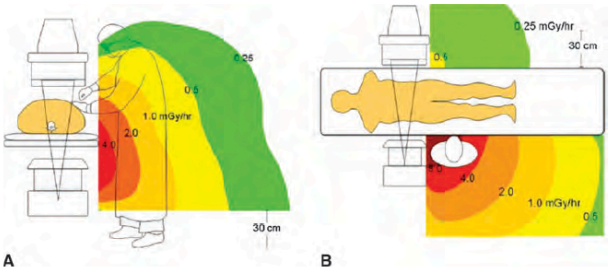
Thicknesses from 0.25 to 0.50 mm are typical. Usually, the lead is in the form of a rubber material to provide flexibility and handling ease. Greater than 90% of the scattered radiation incident on the apron is attenuated by the 0.25-mm thickness at standard x-ray energies.

Radiation safety in interventional radiology

Whenever possible, the less ionizing radiation is used, the better it is for both the staff and the patient. An important potential application of the newer imaging techniques is to develop IR procedures performed without fluoroscopy. Already there are a significant number of IR procedures that can be performed under ultrasound control, and a great deal of work has already gone into developing the equipment and techniques required for guidance using MRI. During CT, radiographic, and fluoroscopic diagnostic procedures, medical staff are seldom in the room with the patient. However, interventionalists and assisting staff are in the room

for most procedures utilizing fluoroscopy, particularly angiographic and IR procedures, and during some CT-guided diagnostic and IR procedures.

This figure below shows the varying intensity of the radiation, primarily scattered x-rays from the patient, during fluoroscopy. Understanding the spatial pattern of radiation can help staff to reduce the doses by where they stand during the procedure and how they position movable shielding. In particular, when the x-ray beam is in a lateral angulation, the stray radiation intensity is much higher on the side of the patient toward the x-ray source than on the side toward the image receptor. The x-ray doses to the heads and arms of staff tend to be least when the x-ray tube is beneath the patient. Several items provide protection for staff during a fluoroscopic or radiographic imaging procedure. The first and foremost is the protective apron worn by all individuals who must work in the room when the x-ray tube is operated.



■ FIGURE 21-16 Dose rates from scattered radiation during fluoroscopy, with the x-ray beam in PA (**A**) and lateral (**B**) orientations. These diagrams show the decrease in dose rate with distance from the location where the x-ray beam enters the patient. When the x-ray tube is beneath the patient (**A**), the highest scatter intensity is to the lower part of the operator's body. When the x-ray beam has a lateral angulation (**B**), the scatter intensity is much less on the side of the patient toward the image receptor. (From: National Council on Radiation Protection and Measurements. *Radiation dose management for fluoroscopically guided interventional medical procedures.* NCRP Report No. 168. National Council on Radiation Protection and Measurements, Bethesda, MD, 2010a.)

For long fluoroscopic procedures, the weight of the apron may become a limiting factor in the ability of the interventionalist and the attending staff to complete the case without substitutions. Some apron designs, such as skirt-vest combinations, place much of the weight on the hips instead of the shoulders. The areas not covered by the apron include the arms, lower legs, the head and neck, and the back (except for wrap- around aprons). During fluoroscopy, the operator commonly looks at the display monitor while x-rays are produced and the x-rays typically strike his or her head from the side and below. Therefore, the protective glasses or goggles should provide shielding on the sides, or the glasses should be of wrap-around designs.
Techniques to minimize radiation exposure include the use of low frame rate pulsed fluoroscopy, lower dose exposure (higher kV, lower mA) and the option of *lastimage-hold*, use of the collimator when necessary, maximizing the source-to-patient distance, minimizing the air gap between the patient and the image intensifier/digital flat panel, and limiting the use of electronic magnifications.

Radiation monitoring: Dosimeters

Practical Aspects of Dosimeter Use

Nearly every medical facility obtains non–self-reading dosimeters, frequently film badges, from a commercial vendor monthly or quarterly. One or more control dosimeters are shipped with each batch. At the beginning of a wear period, typically at the beginning of a month, the new dosimeters are issued to staff and the used dosimeters from the previous wear period are collected. The used dosimeters are returned to the dosimeter vendor for reading. At least one control dosimeter from the same batch is included in the shipment. Control dosimeters are stored in an area away from radiation sources. The vendor subtracts the reading from the control dosimeter from the readings of the dosimeters that were used. An exposure report is received from the vendor in about 2 weeks. However, reporting of unusual exposures or exposures over regulatory limits is expedited. The dosimetry report lists the "shallow" dose, corresponding to the skin dose, the "eye" dose corresponding to the dose to the lens of the eye and the "deep" dose, corresponding to penetrating radiations. Most vendors post dosimetry results on password-secured Web sites.

Placement of Dosimeters on the Body

A dosimeter is typically worn on the part of the torso that is expected to receive the largest radiation exposure or is most sensitive to radiation damage. Most interventionalists, and staff wear a dosimeter at waist or shirt-

pocket level. During fluoroscopy, a dosimeter is typically placed at collar level in front of the lead apron to measure the dose to the thyroid and lens of the eye because most of the body is shielded from exposure. Alternatively, a dosimeter can be placed at the collar level in front of the radiation-protective apron, and a second dosimeter can be worn on the torso underneath the apron.

Technologies

Fluoroscopy

Digital fluoroscopy is a computer-based digital image-processing technique by which real time radiographic images are projected on an image-intensifying fluorescent screen, and in turn converted or digitized for storage or reproduction through an image processor. Compared with conventional fluoroscopy, digital fluoroscopy has several advantages including: post processing that may greatly enhance contrast resolution, high speed image acquisition up to 30 frames/second, and digital image distribution and archiving. The image intensifier of the C-arm unit normally comes in 23 or 30 cm (9 inch or 12 inch) sizes. A C-arm fluoroscopy unit is ideal for most IR procedures. This unit has the advantage of image mobility, permitting various tangential views without moving the patient and allowing patient positioning to facilitate endoscopic or laparoscopic access.

Digital subtraction angiography (DSA)

DSA has become an indispensable tool in angiography and endovascular interventions. DSA refers specifically to techniques by which an initial no-contrast mask image is electronically subtracted from subsequent serial images following injection of contrast medium into the target vessels. After subtraction, the static anatomic structure common to both images is removed; the remaining blood vessels containing contrast medium are opacified. DSA substantially improves the contrast resolution of angiography; however, any slight motion of the structures inside the field of view during the image acquisition may induce remarkable artifacts greatly compromising the image quality.

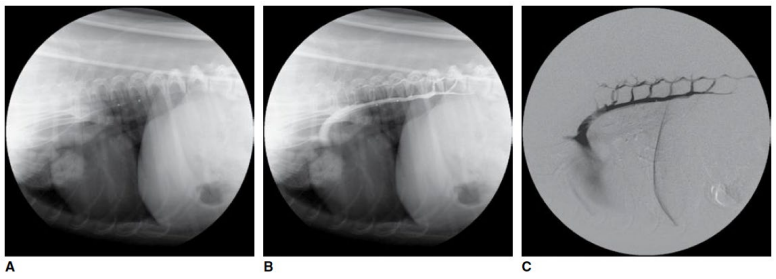
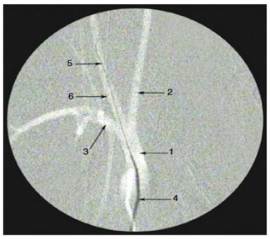


Figure 1.2 Selective digital subtraction angiography of the azygos vein (Lateral view). (A) mask image showing the background structures before injection of contrast medium; (B) The live or contrast image including azygos vein and surrounding anatomic structure; (C) digital subtraction angiogram of the azygos vein with background subtracted.

Roadmapping (also called trace subtract fluoroscopy)

Is the fluoroscopic equivalent of DSA. It is widely used to guide and facilitate endovascular manipulation of the catheter and guide wire. During the procedure, a desired background angiogram, with or without subtraction, is obtained. With the patient remaining perfectly still, the background angiogram is used as a mask to perform subtraction fluoroscopy (roadmapping) in the same field of view. In contrast to DSA, the contrast-filled vessel in roadmapping will appear white, as opposed to black; images of a catheter and guide wire and their motion are visualized superimposed on the background mask image. Roadmapping may improve safety during catheter and guide wire manipulations, reduce radiation exposure and procedure times, and minimize contrast use.



Computed tomography (CT)

The advantage of CT over projection x-ray imaging is that CT is tomographic, presenting the anatomy on a slice-by-slice basis for more exact localization. It is not preferred, at this time, over standard angiographic methods for vascular interventions, because of the difficulty of matching the passage of the bolus through the vascular tree with the acquisition of the appropriate slices. The major disadvantage of CT is that like other x-ray imaging, it uses ionizing radiation, which poses a risk, particularly to the interventionalist and staff. The most widely used interventional CT procedure is for diagnostic biopsy. In addition, CT has been used to guide other percutaneous procedures, to manage fluid collection in the urinary system and to drain abscesses and other fluid accumulations.

Ultrasonography

Ultrasound can be and has been used extensively in IR, particularly for the guidance of biopsy and to aid in fluid management. The major advantages of ultrasound are the lack of use of ionizing radiation, its real-time nature and its low-cost compared to other imaging modalities. The images are tomographic but are not as clear and crisp as those obtained with CT and MRI and may present digestive tract related artifacts, and thus are less acceptable. Ultrasound machines (especially portable) are useful for percutaneous needle access into varies structures (gallbladder, renal pelvis, vessels...) or realization of Doppler studies.

References and images:

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Lecture 7
Gastrointestinal Interventions:
Feeding Tubes & Decompression

Dr. W. Alex Fox-Alvarez, DVM, MS

Objective for the Lecture:

The objective of this lecture is to introduce the student to image-guided gastrointestinal interventions including the percutaneous placement of enterostomy tubes for feeding and decompression.

Objectives:

- Introduce image-guided gastrostomy and jejunostomy
- Introduce image-guided gastrostomy tube for emergency decompression in patients with GDV
- Discuss potential uses in clinical patients

Suggested Reading:

- Weisse, Chick, and Allyson Berent, eds. Veterinary image-guided interventions. John Wiley & Sons, 2015. *Chapter 18: Image-Guided Nutritional Support Techniques.*
- Mack, R. M., Staiger, B., Langlois, D. K., Mehler, S. J., Lam, N., Moore, T., ... & Beal, M. W. (2016). Development and characterization of a technique for percutaneous radiologic gastrojejunostomy tube placement in the dog. Journal of veterinary emergency and critical care, 26(5), 646-653.
- Fox-Alvarez WA, Case JB et al. Temporary percutaneous T-fastener gastropexy and continuous decompressive gastrostomy in dogs with experimentally induced gastric dilatation. *American journal of veterinary research*, *77*(7), 771-778. 2016.

Considerations

Maintenance of an adequate plane of nutrition is critical in the successful management of hospitalized patients. This is particularly true of patients that are behind nutritionally (hypoalbuminemia, cachexia), have large metabolic requirements (burns, trauma), and those with gastrointestinal trauma (surgery, diseases of GI epithelium) in order to restore the normal gastrointestinal barrier. In most cases, nutrition can be provided via nasoesophageal/gastric and esophagostomy tubes without the need for image guidance apart from post-placement radiographs to ensure appropriate placement. However, some patients require placement of a gastrostomy or small intestinal feeding tube to provide long-term enteral nutrition that bypasses the upper gastrointestinal tract. In addition to endoscopic placement, these feeding tubes can also be placed using radiologic image guidance.

Gastrostomy Tubes

Percutaneous Radiologic Gastrostomy (PRG):

Materials: Percutaneous introducer kit (with dilators), Gastric anchors (T-fasteners), Iodinated contrast medium, Gastrostomy tube kit

The gastrostomy tube is placed within the fundus, so the patient is placed in right lateral recumbency. A generous area caudal to the 13th rib is clipped and sterilely prepared. The stomach is insufflated via a nasogastric or orogastric tube and room air until easily palpable. Prior to initiating placement, ultrasound should be used to ensure the spleen is not within the intended site of placement after the stomach is insufflated. Gastric anchors are placed percutaneously in a 3-4cm triangular(3) or square(4) pattern around the desired location of the G-tube. With each anchor placement, appropriate placement of the anchor's deployment needle within the gastric lumen is confirmed via fluoroscopy by injecting 1-3mL of dilute contrast through the needle. Confirmation is indicated when the contrast is seen outlining rugal folds. The placement of these anchors creates a temporart gastropexy to facilitate placement of the G-tube within their area of –pexy. This improves safety and accuracy during placement, and helps maintain a seal around the tube for days to weeks following the procedure as the stoma tract matures. After all anchors are placed and temporary gastropexy achieved, a 5mm skin incision is made at the center of the gastropexy site, and a hemostat is used to bluntly dissect through the subcutaneous tissue to the body wall. Once the body wall is visible, a needle is placed into the gastric lumen within this incision. Successful intraluminal entry is confirmed using positive contrast, as for the anchors, or by withdrawing gas from the lumen. A J-tip guide wire is placed through the needle, into the stomach, and the needle is then removed over the guidewire. Next, the tract sill be dilated over the wire to facilitate G-tube placement. An 11 blade can be used to cut along the guidewire, creating a 2–3 mm stab incision through the body wall, facilitating tract dilation. The tract is serially dilated using well-lubricated dilators until the G-tube can be introduced directly using its stylet, or indirectly through a peel-away sheath large enough to facilitate its placement without a stylet. The G-tube is lubed and advanced through the sheath, over the wire. If a peel-away was used, it is now peeled away for removal. The G-tube balloon is inflated and the tube is pulled outward until the balloon is against the gastric wall. The external bolster is then slid down just above the skin (to allow room for swelling), and sutured to the skin to maintain position. The guide wire is finally removed. Placement is confirmed via fluoroscopy and injection of a small volume of contrast through the gastrostomy tube.

Percutaneous Radiologic Gastrojejunostomy (PRGJ)[1]:

Materials: Percutaneous introducer kit (with dilators and Berenstein catheter 4/5F 65 cm), Gastric anchors (T-fasteners), Iodinated contrast medium, stiff straight-tip hydrophilic guidewire (.035, 150 cm), Berenstein catheter (included in most percutaneous, GJ feeding tube

For a PRGJ, you do the same as you would for the PRG tube placement, however after placing the needle though the body wall into the gastric lumen, a stiff straight-tip hydrophilic guidewire (.035, 150 cm) is introduced through the needle instead of the j-tip guidewire. Under fluoroscopic guidance, the guidewire is maneuvered through the pylorus, duodenum, and into the jejunum. If difficulty is encountered navigating through the pylorus, sometimes altering the patient's position can facilitate placement. Following the guidewire reaching the jejunum, the tract is dilated until a peel-away sheath can be placed. The GJ-Tube is advanced over the wire, and 3-5mL of dilute contrast is injected under fluoro to confirm appropriate placement. The wire is removed, and the tube is secured as described for the PRG.

Gastrostomy tube for decompression[2]:

For sustained gastric decompression in dogs with GDV, the following has been described. The gastrostomy tube is placed within the fundus, however the fundus is more accessible on the right side of the body due to the torsion, so the patient is placed in left lateral recumbency. A generous area caudal to the 13th rib is clipped and sterilely prepared. Under ultrasound guidance, three gastric anchors (T-fasteners) are placed percutaneously about 1.5cm apart in a triangular orientation, into the gastric lumen via the right dorso-lateral abdomen. The exact location of placement within this region is determined via ultrasound and the degree of palpable tympany. Prior to placement, all T-fasteners are attached to a 6cc syringe with 2cc sterile saline. After introduction into the gastric lumen, the syringe plunger is drawn back to evaluate for the presence of air bubbles, indicating appropriate intra-gastric positioning, and the gastric anchors are deployed from the needle. In the center of the temporary gastropexy, a 21ga needle is placed into the gastric lumen, into which a 0.018in, 60cm guidewire is fed. The needle is removed over the guidewire and a 5fr introducer is placed over the guidewire to dilate the tract and is then removed. Lastly, a 5fr locking pigtail catheter (Dawson Mueller Catheter) is placed over the guidewire into the gastric lumen, the guidewire is removed and the pigtail coil is formed, locked in place and retracted to the gastric wall. Intermittent flushing of the decompressive G-cath with 3-5mL of saline followed by 3-5mL of air is recommended every 10-15 minutes to ensure patency is maintained. The gastric anchors and G-cath are removed at the time of surgical explore, *before* derotation is attempted.

Stoma Care

Gastrostomy tubes must remain in place for a minimum of 4 weeks before the tube is removed to ensure mature stoma tract formation has occurred. Consider a longer duration in patients on corticosteroids or with severe chronic illness. Gastric anchors should not be placed too tight to prevent erosion of the gastric mucosa, or dermal pressure sores under their external bumpers. Gastric anchors can be left in place for the first 1-4 weeks until at least a fibrous tract has formed. If the tube is dislodged prematurely, the gastric anchors will decrease the risk of abdominal spillage, and facilitate replacement of the feeding tube before a mature tract forms. The replacement tube's location should be confirmed radiographically. Once a mature tract has formed, gastric anchors can be released and removed.

Tube length and location should be monitored daily for evidence of migration. Stoma sites should be evaluated daily for redness, dwelling, discharge, and pain. Daily primary layer dressing changes are recommended for the first 3-5 days. Early stoma site infections are common, and are often easily treated with topical and/or systemic antibiotics. If granulation tissue develops around the stoma site over time, it may be cauterized using silver nitrate sticks to minimize bleeding when manipulated.

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Lecture 8

Intrahepatic portosystemic shunts; transvenous and fluoroscopy-assisted surgery

J. Brad Case, DVM, MS, DACVS
ACVS Founding Fellow, Minimally Invasive Surgery
Associate Professor, College of Veterinary Medicine
University of Florida, Gainesville, Florida, USA

OBJECTIVES

- **To know the vascular anatomy of the liver and IHPSS in dogs and cats**
- **To understand the principles and procedures of PTCE**
- **To understand the clinical benefits and outcomes of PTCE**

Intrahepatic Portosystemic Shunts

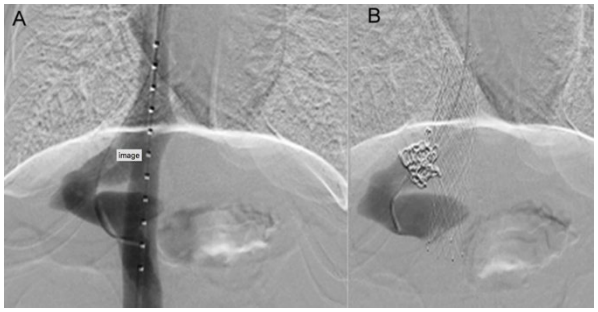
Congenital intrahepatic portosystemic shunts (CIHPSS) are vascular anomalies which divert portal blood away from the liver. Vascular deprivation leads to hepatic underdevelopment, insufficiency and in many cases, liver failure. Hepatic failure can be associated with many morbid clinical consequences including: seizures, ataxia, obtundation, anorexia, vomiting, diarrhea, pollakiuria, stranguria and death.[1-3] Consequently, surgical intervention aimed at perfusion of the starved liver is recommended.[4-10] To date, a number of surgical techniques have been recommended to correct perfusion of the liver in dogs with CIHPSS. Surgical techniques can be divided into *extravascular* and *intravascular* and can result in either *partial* or *complete* attenuation of the aberrent portal blood flow. Gradual occlusion has been performed most frequently because complete occlusion of CIHPSS is rarely feasible.[4-15] Partial suture ligation has been reported most commonly in veterinary medicine.[4,5,11,12,16] Partial suture ligation causes sudden, nonprogressive attenuation, and persistent vascular shunting which may lead to persistence of clinical signs and a poor outcome in some dogs. Perioperative complications range from 40-77%, perioperative mortality ranges from 13-27% and median survivals were between three and four years.[4,5,11,12,16] To overcome the limitation of static attenuation, gradual occlusion devices have been utilized to eliminate persistent shunting in dogs with CIHPSS. Ameroid constrictors, cellophane bands and hydraulic occluders have been used clinically and they cause progressive gradual attenuation.[6-8] Clinical results were improved when compared to previous reports with perioperative complications ranging from 9-55%, perioperative mortality ranging from 0-27% and 2-year survival between 60-80%. [6-8]

More recently, the results of intravascular, percutaneous, transvenous, coil embolization (PTCE) for CIHPSS attenuation in dogs have been reported with improved clinical outcomes.[17-20] In these series, perioperative complications ranged from 8-16%, perioperative mortality was 5-8% and median survivals were > 6 years.[17,19,20] In one recent study comparing outcomes between PTCE and cellophane banding, postoperative complications were significantly more frequent and hospitalization time was 3 times longer in the cellophane banding dogs.[19]

Percutaneous Transvenous Coil Embolization

A summary of the PTCE procedure is described here. However, the reader is referred to a

complete textbook on the procedure and instrumentation required before attempting in client-owned dogs.[18] Dogs are positioned in dorsal recumbency (left and right division IHPSS) or left lateral recumbency (central division IHPSS) for PTCE. The cervical region is clipped and prepared using aseptic technique. The Seldinger method is used to place an 11 or 12 Fr. introducer sheath in the right external jugular vein (EJV). Fluoroscopy is used to guide placement and position of the introducer. A 4 French, 65 cm, 0.035 Angiographic (Berenstein/Kern) catheter is then used to select the shunt and a 5 French, 65 cm, 0.035 Angiographic (Marker) catheter is placed along side the 4 Fr. catheter into the caudal vena cava for calibration. Resting caudal vena cava and portal pressures are measured using a transducer. A manual breath-hold maneuver is performed by the anesthetist and a combined digital subtraction porto-cavogram is performed using 6-8 mls of iodinated contrast diluted 1:1 in sterile saline and measurements made/confirmed for stent and coil selection. An 0.035, 260 cm, straight tip exchange length Guide wire is advanced into the caudal vena cava via the marker catheter and the marker catheter is removed. Next, a self-expanding, laser-cut Nitinol stent (Vet-stent Cava) is advanced over the guide wire and deployed with the dog temporarily paralyzed. The stent is placed to maximize coverage of the portosystemic shunt ostium (minimum of 2 cm on either side) and to maintain adequate distance from the right atrium of the heart and right renal vein. Following stent placement, a 4 French, 100 cm, 0.038 Angiographic Kerns catheter is advanced over the guide wire and the guide wire removed. The Kerns catheter is used to select the portosystemic shunt across the stent and beyond the level of the caval ostium. A 0.035, 150 cm, hydrophilic glide wire is used to guide the catheter progressively into the shunt. The pressure transducer is attached to the Kerns catheter for continuous portal pressure monitoring. A Cobra-style angiographic catheter is advanced along side the Kerns catheter and used to select the shunt across the stent just inside the caval ostium. Careful attention to the size and length of the shunt ostium is critical to select and place coils safely, especially in smaller dogs and cats. Digital subtraction angiography is performed to confirm appropriate position within the shunt. Next, 0.035 platinum coils are deployed into the shunt via the Cobra catheter immediately adjacent to the vena caval stent. The size and shape (cylindrical versus tornado) of the coils depends on the size and morphology of the portosystemic shunt which in small breeds tends to be smaller with a narrower landing zone. **Figure 1** Combined portocavogram (A) and post-PTCE angiogram (B) in a dog with a right IHPSS.[19] The current recommendations are to increase the portal-caval pressure gradient by 5-6mmHg. Once the target portal pressure increase is reached, a final digital subtraction angiogram is performed and a plain radiograph obtained to document implant position. The introducer sheath is removed and digital pressure (with or without an interrupted stitch in the vessel wall) is applied for 25 minutes to the venipuncture site for hemostasis.



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Lecture 9

Arteriovenous Malformations: fluoroscopy-assisted and transarterial surgery

J. Brad Case, DVM, MS, DACVS
ACVS Founding Fellow, Minimally Invasive Surgery
Associate Professor, College of Veterinary Medicine
University of Florida, Gainesville, Florida, USA

OBJECTIVES

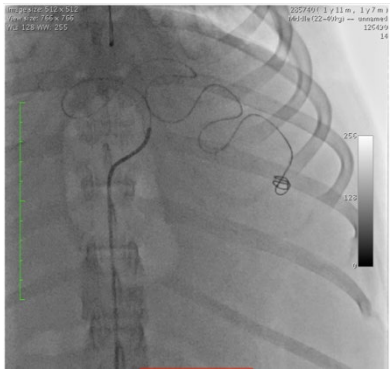
- **To understand the complex vascular anatomy and pathophysiology of hepatic arteriovenous malformations (HAVM)**
- **To understand the principles of angiographic assessment and interventional treatment**
- **To understand the prognoses and clinical sequela of successful embolization of a HAVM**

Hepatic arteriovenous malformations

Congenital hepatic arteriovenous malformations (HAVM) are rare in dogs and are associated with significant complications and mortality.[1,2] Additionally, high-flow HAVM are risky to resect surgically and are challenging to completely embolize.[2,3] However, successful management of dogs with HAVM can be accomplished using advanced imaging and interventional coil and glue embolization to completely eliminate the main fistula and supplying nidus.[1,4] Hepatic arteriovenous malformations are aberrant connections between the hepatic arteries and portal veins. HAVM can range in shape, size and number or type of vascular anastomosis. The Cho-Do classification system is used in human medicine to predict prognosis and treatment success based on the type of AVM in question.[3] The veterinary literature is limited to 4- and 1- dogs undergoing glue embolization alone or in combination with coils for treatment of their hepatic arteriovenous fistulae.[1,4] In these reports, all dogs survived the procedure and were alive between 6 and 17 months following treatment. One operative complication occurred in which glue embolization of the distal aorta caused temporary loss of pulse but no long-standing clinical problems resulted.[1] Clinical outcome in these dogs was good or fair in four- and poor in one-dog who required surgery after persistent shunting was documented at follow up.[1,4] In comparison to dogs undergoing surgical lobectomy, dogs treated by glue embolization had reduced perioperative mortality and an improved clinical outcome in one study.[1] A dog recently treated at the University of Florida Small Animal Hospital possessed characteristics of a Cho-Do type 1 AVM (direct communications between the left hepatic artery and portal vein); and a Cho-Do type 3b with left hepatic arteries-arterioles forming aberrant anastomoses with high-output hepatic veins. Treatment options considered were complete surgical resection of the left hepatic division or complete embolization of the main arteriovenous communication as well as the associated nidus. The dog underwent complete endovascular embolization and had a rapid clinical response including resolution of abdominal discomfort and ascites within a few days. Follow-up examination and imaging at 8 weeks post embolization revealed complete embolization on CTA in addition to hepatic artery dilation (compensatory blood flow) and a large portal vein thrombus due to chronic portal venous hypertension and hepatofugal blood flow.[4]

Percutaneous transarterial arteriovenous embolization

The patient is placed in dorsal recumbency and the procedure is performed via the femoral artery after a cut down and introducer access using the Seldinger technique. A 5 Fr. Introducer is typically used. A 5 Fr. reverse curve angiographic catheter over an 0.038" hydrophilic glidewire is used to access the celiac artery. A celiac arteriogram is then performed. Next a 3 Fr. Microcatheter is used to select the branch of the hepatic artery supplying the HAVM via the reverse curve support catheter (**Figure 1**). A diagnostic angiogram is then performed to determine the morphology and flow characteristics of the HAVM. Once the HAVM is identified, depending on the flow dynamics and regional vasculature embolization can be considered. In some cases, flow may be too great for glue embolization alone. In such instances, flow can be attenuated or reduced using permanent solid-state embolics such as 0.018" platinum microcoils (**Figure 1**). Once flow is adequately reduced to minimize the risk of non-target embolization, liquid embolics such as glue (N-butyl Cyanoacrylate) can be used to complete the embolization including all subsegmental *parasitizing* vessels supplying the nidus. Glue embolization is performed in combination with nonionic radio-opaque substances such as Lipiodol to facilitate angiographic visualization. A final celiac arteriogram is performed to confirm the absence of remaining arterial communications (nidus) with the AVM. Although vascular closure devices are available, they are expensive and can be challenging to use. Femoral artery ligation is reasonable and is well tolerated by dogs. Prolonged digital pressure or primary repair of the vessel are also possible.



Hybrid transarterial arteriovenous embolization

The patient is aseptically prepared including the abdomen, inguinal region and medial thigh; and positioned in dorsal recumbency on a radiolucent operating table. The abdomen including the inguinal and proximal thigh is draped. A cut down to the femoral artery or ultrasound-guided introduction of a 5 Fr. introducer into the femoral artery is performed. Briefly, an 18-gauge catheter is inserted into the femoral artery, which is then exchanged for a 4 Fr. microintroducer over an 0.018" guidewire. A 5 Fr. introducer/sheath is then used to replace the microintroducer over an 0.035" guidewire. A ventral midline celiotomy is then created to expose the gastrointestinal tract and liver. Abdominal exploratory typically reveals ascites, a small liver, as well as multiple tortuous aberrant vessels within the cranial abdomen. A jejunal vein is located and catheterized with a 22-24 gauge catheter to allow for direct continuous portal pressure monitoring. Significant portal hypertension is to be expected due to arterial inflow to the portal system.

Most operating room C-arms are adequate for angiographic assessment although newer high-resolution fluoroscopy units with digital subtraction angiography are preferred. A 4 F.r Berenstein angiographic catheter is then placed in the femoral introducer and advanced into the celiac artery (confirmed via fluoroscopy). The catheter is then advanced into the common hepatic artery. Hepatic artery angiography is then performed with a manual injection of a 1:1 iohexol and sterile saline solution. No more than 3-5mls

is required for a complete study. In general, the least amount of contrast that is needed to complete the procedure is used. However, 3-5 mls/kg appears to be safe and unlikely to results in contrast-related complications such as kidney injury. Typically, one-to-two major aberrant arteries/arterioles will be noted branching from the major hepatic artery, feeding into a large, tortuous nidus, which then decompresses into the portal vein in most cases. Hepatofugal blood flow is often documented along with multiple acquired shunts. Next a 3 Fr. Microcatheter is used to select the branch of the hepatic artery supplying the HAVM via the Berenstein support catheter. 0.018" MREye microcoils are placed in the aberrant vessels branching off of the hepatic artery and feeding into the nidus. A decrease in AV flow should be noted and portal pressure may or may not decrease significantly depending on how much collateral arterial supply exists. In many HAVM one-to-two dominant outflow veins will be present and can be ligated with suture or hemoclips. In many cases, cessation of AV shunting and a significant drop in portal pressure will be observed. Repeat arteriography should confirm complete attenuation of AV flow and no evidence of new feeding arteriolar vessels. In some cases, the ipsilateral phrenic artery or other smaller hepatic arteriole branch will be observed parasitizing the AVM. In order to provide complete attenuation, glue embolization will be required to completely eliminate the nidus. A microcatheter, primed with a sterile 5% dextrose solution, is advanced through the support catheter (over a 0.018" guidewire) and a 1:1 to 1:3 mixture of n-butyl cyanoacrylate/Lipiodol (in a primed syringe) is delivered into the nidus and including the major feeding arteries/arterioles. During delivery, the microcatheter is slowly retracted and removed before the glue catalyzes and locks the catheter in the hepatic artery. Complete embolization is confirmed via arteriography and the end portal pressure documented. The jejunal vein is ligated proximal to the catheter site to facilitate catheter removal. The body wall, subcutaneous tissue and skin were closed routinely. The femoral artery is repaired or ligated to allow for closure of the arterial cut-down site.

Outcome and monitoring

Although the literature is limited, dogs undergoing interventional management of their HAVMs appear to have an excellent short-term outcome and a fair-to-good long-term outcome. Clinical improvement is common and likely relates to resolution of portal hypertension, visceral congestion and ascites. Visceral congestion and ascites is known to cause discomfort and increase bacterial translocation and end-organ deposition of bacteria and endotoxins which in turn worsens hepatic function and encephalopathy. Additionally, bacterial overgrowth and intestinal malabsorption can be seen with chronic portal hypertension. Dogs with HAVM have underdeveloped livers with acquired portosystemic shunts which do not resolve with successful treatment in most cases. Consequently, monitoring and on-going medical management of liver insufficiency and encephalopathy should be anticipated. Interestingly, portal vein thrombosis appears to be common following successful embolization of HAVM. Despite this seemingly severe occurrence most dogs appear to tolerate the lack of hepatopetal portal blood flow well. This is likely due to the presence of acquired portosystemic shunts and the arterial perfusion present in the liver. Consequently, it should be considered paramount to maintain as much hepatic arterial flow to the remaining liver as is possible. In some cases, incomplete or recurrent HAVM shunting results after treatment. Repeat treatment after diagnostic angiography is considered in these cases and is common in human

medicine. On-going monitoring of hepatic function should include: recheck examinations at 1 month and every 4-6 months lifelong. Blood work should include: CBC, serum chemistry, urine analysis and Protein C. Ammonia levels can also be monitored if encephalopathy is a concern.

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Lecture 10
Interventional Cardiac Procedures

Pacemaker implantation, pulmonic balloon valvuloplasty (BVP) and PDA occlusion are the most common minimally invasive cardiovascular procedures. However, other procedures are less commonly performed such as cardioversion of horses with atrial fibrillation, arterial occlusion for epistaxis in horses, AV fistula occlusion, septal occlusion and balloon dilation of cor triatriata.

Pacemaker implantation

Pacemakers are implanted to treat symptomatic bradycardias typically caused by 3rd AV block, 2nd AV block and sick sinus syndrome. The patients should have further investigation to look for intercurrent diseases but the extent of these may depend on the severity of the clinical signs. A troponin assay can help evaluate potential myocarditis which could indicate a poorer prognosis. In small dogs and cats, an epicardial lead should be used with the generator placed in the abdomen.

Temporary pacing is essential and this can be achieved by external pacing patches or a temporary pacing lead which is introduced via a saphenous or jugular vein under sedation. The lead is guided fluoroscopically into the right ventricle and attached to a temporary pacing box. Once this has been achieved, the patient can be anesthetized for permanent pacemaker implantation.

The right jugular vein is exteriorized and stabilized between vascular loops. A stab incision is made and the pacing lead introduced. A vein pick can help. The lead is then advanced into the right atrium. At this point, the stiffening wire should be removed and curved to 60 – 80' which will facilitate directing the lead into the right ventricle when replaced in the lead. The lead should be advanced into the right ventricle. Two different leads are available. Passive fixation leads have tined ends that catch in the trabeculae of the right ventricle. Active fixation leads have a screw ending which is screwed into the RV myocardium using a wrench provided. The stability of these leads is determined by gentle traction unlike the passive leads. Once placed the lead impedance and thresholds can be checked using the interrogator. Lead impedance is usually 400 – 1200 Ohms.

The generator is implanted dorsally over the right neck and the lead tunneled through the subcuticular tissue. Connection is made using a screwdriver provided ensuing the lead in pushed all the way into the generator. The screw has been adequately tightened when clicking can heard with rotations. The tissue is then closed in a routine manner and a light pressure bandage applied to the neck. The bandage can be removed after 7 days if there is no seroma formation. The patient should have leash exercise for one month using a harness before a gradual return to exercise. This allows time for the lead to form a firm attachment to the endomyocardium. A loose collar can be worn but a leash should not be attached due to the risk of lead fracture.

Major complication include lead dislodgement and infection. The patient should return after one month for pacemaker interrogation and reprogramming as fibrosis at the lead tip can alter required depolarization settings. This is followed by annual rechecks.

Balloon valvuloplasty for pulmonic stenosis

BVP is indicated for cases of severe pulmonic stenosis. Severity is graded by the systolic pressure gradient across the stenosis as measured by echocardiography with mild being 20 – 50 mmHg, moderate 50 – 80 and severe over 80 mmHg. Studies have shown an increase in survival in dogs that experienced a significant (over 50%) reduction in pressure gradient. In pulmonic stenosis, the valve leaflets may be normal but fused (type A) or thickened and dysplastic (Type B) or a combination (Intermediate). Type A valve have a better long term outcome as the leaflets tear during ballooning.

Patients are usually treated with a beta blocker for a minimum of 2 weeks prior to the procedure to reduce the dynamic obstruction and risk of arrhythmias. It may also mitigate the risk of a "suicide" right ventricle post ballooning. Some cardiologist give the class 1 anti-arrhythmic procainamide at induction but lidocaine should be available as ventricular arrhythmias are common. The approach can be via the femoral or jugular vein depending on the size of the patient and size of the right atrium. Either a venous cutdown can be used or a percutaneous approach using a modified Seldinger technique. Initially, a BWP catheter is introduced into the pulmonary artery and the pressures in the PA, RV, and RA measured via a pull back technique. A multi-ended catheter such a pig-tail or NIH ids advanced into the RV outflow tract and used for contrast injection. This is used to assess the size of the pulmonary annulus and look for normal coronary artery anatomy as an R2A anomaly would be a contraindication. A balloon is chosen that is 1.2 – 1.5 times the diameter of the pulmonary annulus.

The BWP catheter is advanced into the PA and a guidewire introduced. Stiffer wires provide more stability but are less well tolerated. The wire is less likely to back into the RV if it is far in the lung lobes. The BWP catheter is withdrawn leaving the wire in place. This is used to guide the balloon into position. It is important that the wire on the table is flat and straight so it acts like a train track the catheters and balloons move along. Once in position, the balloon is inflated several times to the stated burst pressure with time allowed for the cardiovascular parameters to recover. The balloon is withdrawn and the BWP catheter re-introduced to measure pressures as before. Closure is routine for a cutdown with the vessel sacrificed. With percutaneous access, pressure is maintained for 30 minutes.

The patient can usually be discharged the following day and maintained on beta blockers. The pressures are re-evaluated at 3 months and the beta blockers may be discontinued if they are mild to low moderate. In dogs with higher gradients, annual rechecks are recommended as the high gradients can recur.

PDA Occlusion

Any dog with a continuous machinery murmur should be evaluated as soon as possible as the sooner the PDA is closed, the less the left heart will be volume loaded. An apical systolic murmur of secondary mitral regurgitation may be detected. The choice of catheter v surgical occlusion is based on the size of the patient and ductus, cost and owner preference. A large ductus in a small patient may require surgical closure as the vessels may be too small for the catheters required. The diagnosis is confirmed by echocardiography. Cats with PDAs often have systolic murmur and high PA pressures. Under general anesthesia, the patient is placed in right lateral recumbency with the left hindlimb elevated to reveal the right groin. The femoral artery is dissected out and bathed in lidocaine to minimize the risk of vasoconstriction. The artery is entered using a Seldinger technique and a large catheter such as a CheckFlo introduced into the artery

along the exchange guidewire. This is advanced to the level of the ductus and contrast injected as previously described. This is used to visualize the ductus and size the closing device. Recent studies with 3D transesophageal echo suggest the ductus may be oval in some cases which could result in incorrect sizing. Coils are used less commonly as they are more likely to embolize into pulmonary artery and leave more residual flow. The Amplatz canine duct occlude (ACDO) is the most frequently used device.

The catheter is advanced across the ductus into the pulmonary artery. Atropine should be available in case the Branham effect occurs. The ACDO is prepared by exteriorizing it into a bowl of saline (not heparinized) and then making sure it is slightly loose on the wire. It is then withdrawn into the delivery catheter which is pushed up against the CheckFlo and advanced into the larger catheter and along its lumen until the first disc is exteriorized in the pulmonary artery. The ACDO and catheter are withdrawn until tension is felt as the ACDO is pulled up against the PA ductus orifice. While tension is maintained, the catheter is withdrawn allowing the second disc to exteriorize in the ductus. Stability is tested by a gentle push-pull on the guidewire. The ACDO is left for 10 minutes in situ and a low velocity contrast injection made to confirm ductal closure. The wire is then detached from the ACDO by turning. Finally lateral and DV radiographs are taken to confirm position.

The femoral artery is ligated and closure is routine. Attempts at percutaneous access have not been successful as hemorrhage post operatively can be fatal. Similar to pacing, exercise is restricted for the first 4 weeks while the ACDO embeds in the tissues. Dislodgement is rare after this time. Device infection is also rare.

Summary

Minimally invasive treatments for cardiovascular diseases are a developing field as new devices and techniques are developed. They have the potential to avoid the need for bypass surgery and this field will continue to expand in the future. Success rates are not widely published but at UF, or major complication rate is about 2.5%.

Lecture 11

Fluoroscopic-assisted ureteral surgery: ureteral obstructions

Penny J. Regier, DVM, MS, DACVS-SA

Department of Small Animal Clinical Sciences, University of Florida

Objectives

1. Be able to identify and discuss indications and contraindications for ureteral reimplantation
2. Be able to identify and discuss indications and contraindications for ureteral stents
3. Be able to list goals of ureteral stent placement.
4. Be able to identify and discuss indications and contraindications for subcutaneous ureteral bypass (SUB)
5. Be able to list complications and complication rates for each surgical procedure.

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Ureteral Obstruction

The longer the duration of ureteral obstruction, the less likely that the kidney will recover to where the animal is no longer azotemic. In normal dogs that have suffered 1 week of obstruction GFR returns to 65% of control values after the ureter is unobstructed. Maximum recovery occurs by ~5 weeks. In normal dogs in which the obstruction is removed after 2 weeks, GFR returns to only 46% of normal over 4 months. Slight, moderate, and severe tubular dilatation with interstitial fibrosis occurs after 1, 2, and 3 weeks of obstruction.

Whether this impedes ureterolith migration, resulting in secondary ureteral injury, is unknown. Approximately 75% of cats have a unilateral obstruction.[18] The left feline ureter often has a bend in its course ~ 3cm distal to the kidney. This seems to be a common location for obstruction by ureteroliths. The presence of a right circumcaval, or retrocaval, ureter is not uncommon in cats. ~10-15% of cats with right-sided ureteroliths may have a retrocaval ureter on that side.

Feline Ureterolithiasis

Obstructive ureterolithiasis is one of the most common reasons for performing ureteral surgery, especially in cats. After ureteral obstruction has been identified, the patient should be stabilized before surgery, especially if dehydrated. The benefits of potentially avoiding surgery must be weighed against the risks for increased renal damage secondary to prolonged obstruction. Kyles et al. reported a study of 153 cats, 52 cats of which were treated by medical management alone; 9 of 14 that had serial imaging performed had migration of an ureterolith into the bladder. An additional 101 cats in the study required surgery for ureteroliths that did not migrate with medical management.[1] Of the 52 cats in which medical management alone was attempted, 17 were euthanized or died, 12 were lots to follow-up, 16 had no change in creatinine concentration, and only 7 had a significant improvement in creatinine concentration with medical management alone. One- and 2-year survival statistics were 66% and 55%, respectively, compared with 91% and 88% for those treated surgically.[1]

Presurgical Considerations

Every cat has a different amount of damage to the obstructed kidney, and the clinician cannot predict how long the obstruction has been present or how well the cat will rebound. We are unable to predict if the cat's creatinine concentration will normalize or decrease slightly or not improve. If an azotemic cat has unilateral ureteral obstruction, it must have bilateral renal disease; otherwise, the cat would not be azotemic or uremic. Fixing the obstruction does not take care of the underlying renal disease. Significant complications occurred in 1/3 of feline cases that were taken to surgery to remove ureteroliths(s), with overall mortality rate reported to be 18-21%.[100] The most common complication was development of uroabdomen (6-16% of cases).[100] Uroabdomen may occur secondary to leakage at the ureterotomy or ureteral reimplantation site or from the greater curvature of the kidney at the nephrostomy tube site. Newer nephrostomy tubes designed for percutaneous placement may result in less risk for leakage and have been used successfully in the preoperative and postoperative periods for renal decompression.[6]

Localizing the Ureterolith

Localization of a ureteral obstruction is most commonly performed using abdominal ultrasonography. Ultrasound allows visualization of calcium oxalate, small, and radiolucent ureteroliths and is also able to determine the degree of ureteral and renal pelvic dilatation present. If mineralized calculi are not evident on ultrasonographic examination, ureteral dilatation or hydronephrosis may be secondary to a ureteral stricture from a previous calculus, or a blood calculus may be present. Hematuria appears to be commonly seen on urinalysis of cats with obstructive blood calculi. Excretory urography can also be performed, but image quality may suffer in animals that are azotemic, and there is some risk for inducing further renal damage if the patient is not adequately hydrated.

Surgery

After surgery is deemed necessary, a ureteral resection with reimplantation or ureterotomy is performed. Alternatively, placement of double-pigtail ureteral stents

under fluoroscopic guidance may be performed. In most cases when a stent is placed, an ureterotomy is not performed, thus eliminating the risk for postoperative leakage at this site. More recently, SUB has been reported, which allows urine to bypass the ureter altogether via a nephrostomy tube, to a subcutaneous injection port, and then to a cystotomy catheter.

Neoureterocystotomy/Ureterotomy

Ureterotomy or neoureterocystotomy in small dogs and cats required magnification of the surgical site with either head loupes or an operating microscope. Microvascular instruments and sutures (7-0 to 10=0) are required. All procedures are performed via a standard ventral midline celiotomy. Ureteral reimplantation (neoureterocystotomy) is recommended for mid to distal ureter obstruction. The surgical procedure may also be performed for proximal obstructions in conjunction with a nephrocystopexy or renal descensus and psoas cystopexy. In cases with distal ureterolithiasis, this is most commonly performed if there is significant fibrosis identified at the site of obstruction and ureteral patency is going to be questionable after ureterotomy. The ureter is ligated at the level of the bladder, and any ureteral segment containing a calculus or mass is resected. The distal end of the dilated section of ureter is sutured to the urinary bladder mucosa near the apex with an intravesical technique, which requires a ventral cystotomy, or an extravesical technique. Kyles et al.[1] reported a 40% recurrence of ureteral obstruction from stone formation or passage of a previous nephrolith after ureteral reimplantation. An ureterotomy may be performed in the case of a single obstructive stone but is not ideal in the case of multiple stones, nephroliths, and strictures.

Ureteral Stent

The goals of ureteral stent placement include the following: urine diversion, passive ureteral dilation, decrease surgical tension, and prevention of nephrolith migration/ureteral obstruction. Although retrograde stent placement can be performed with the aid of a cystoscope in female cats (nonsurgical), this is less successful (4 of 21 (19%) of female cats attempted) than surgical placement through a ventral midline celiotomy. A guidewire is placed, either retrograde from the ureteral orifice to the renal pelvis or normograde (preferred) through the greater curvature of the kidney, down the ureter, past the ureterolith, and into the bladder. A ureteral dilator is then passed over this wire, followed by placement of a double-pigtail indwelling catheter. One end of the catheter lies within the renal pelvis and the other in the bladder lumen. The ureterolith(s) are not typically removed, but can be, via a ureterotomy, if there is difficulty passing the guidewire around the obstruction. Reported complications include stranguria/pollakiuria, presumably due to irritation of the trigonal region from the distal stent; imperfect stent location; ureteral trauma during stent placement; and urinary tract infection.[7,17] Most complications associated with stents are reported to be minor; however, stent removal or replacement was required in 19 of 70 (27%) cats because of long-term complications, including stent occlusion and dysuria.[3,7] In another study, stent obstruction secondary to encrustation was documented in 3 of 4 cats in which a ureteral resection-anastomosis was also performed.[17] Stricture formation at the surgical site may have contributed to obstruction in these cases.

Berent et al.[3] reported the outcomes of ureteral stenting in 69 cats with benign ureteral obstruction. This study reported a 95% success rate with stent placement and also reported that short and long-term complications were typically minor but may require stent exchange in 27% of cases. Peri-op mortality rates (7.5%) were improved compared to traditional surgical options (21%).[3] Wormser et al.[4] evaluated 117 cases of ureteral surgery and ureteral stenting and found a 22% rate of ureteral reobstruction and a chronic lower UTI rate of 11%, which was more common in cats that underwent stent placement. These cases had similar MST and periop mortality rates. Of the cases with follow-up, 20% (37% with stent and 2% without stent) had chronic hematuria, stranguria, or dysuria following ureteral surgery and an additional 14% of cats had chronic UTIs.[4] Culp et al.[5] reported that cats with ureteral stents had significantly greater decreases in BUN/creat concentration 1 day after surgery and at hospital discharge compared to cats with ureterotomy; cats with stents were significantly more likely to have resolution of azotemia prior to discharge.

Subcutaneous Ureteral Bypass (SUB)

A SUB device (Norfolk Vet Products) consists of locking-loop nephrostomy and cystotomy catheters connected under the skin via a specialized port. During laparotomy the nephrostomy catheter is placed under fluoroscopic guidance using a modified Seldinger technique: a guidewire is passed through an 18 gauge catheter into the renal pelvis; after catheter removal the nephrostomy catheter is passed over the guidewire into the renal pelvis. The tube is secured in place by locking the pigtail and by placement of sutures or sterile tissue glue between the tube's fenestrated cuff and the renal capsule. The cystotomy tube is placed surgically into the bladder through an apical purse-string suture, and the catheter cuff and overlying silicone ring are secured en masse to the serosal surface of the bladder with sutures and tissue glue. The abdominal muscle on the side ipsilateral to the nephrostomy tube is exposed through a skin incision lateral to the laparotomy incision. The free ends of both catheters are tunneled gently through the abdominal wall and directed toward the port, with the nephrostomy catheter end toward the caudal barb and the cystotomy catheter end toward the cranial barb. Such positioning allows both catheters to maintain a gently curving transabdominal course in an effort to prevent kinking. The strings of the locking loops are kept tight with hemostats and trimmed short, and the catheter ends are advanced over the appropriate barbs. The string end and catheter-barb junction on each side are covered with a boot, and the port is sutured to the body wall with nonabsorbable monofilament suture. More detailed instructions are available online.

Urine collection and fluid infusions through the SUB port are performed with a 22 gauge Huber needle to prevent damage to the silicone diaphragm. Flushing with sterile saline every 3-6 months is recommended to ensure patency and reduce encrustation. With this precaution, 92% of systems remain patent long-term (median, 18 months).[2] Patency can be confirmed by flushing under ultrasound guidance or with fluoroscopic pyelography using an infusion of a 1:1 iohexol-saline mixture. Over-distention of the renal pelvis must be avoided during flushing. Complications of the SUB device include occlusion from urolith material (~13%) or blood clots (<3%), kinking (3%), and rarely urine leakage.[15] Deroy et al.[14] recently compared double-pigtail ureteral stents and SUB devices for treatment of ureterolithiasis in cats and found the following: 1) median

duration of surgery and hospitalization longer in the stent group, 2) periop mortality rate of 18% in the stent group vs 13% with SUBS, 3) shorter MST in the stent group, 4) greater risk of LUT-related signs with stent placement, 5) higher rate of occlusion with stents (26% vs 4%), and 6) cats requiring additional procedures to treat complications was 44% with stents vs 9% with SUBS.

Lecture 13

Lymphatic interventions: chylothorax

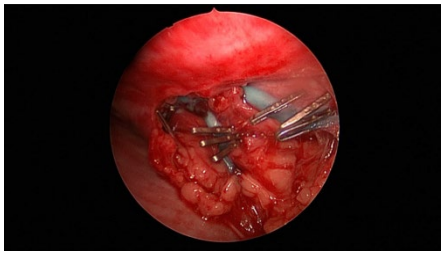
J. Brad Case, DVM, MS, DACVS
ACVS Founding Fellow, Minimally Invasive Surgery
Associate Professor, College of Veterinary Medicine
University of Florida, Gainesville, Florida, USA

OBJECTIVES

- **To understand the lymphatic anatomy and pathophysiology of chylothorax**
- **To understand the principles of lymphangiographic assessment and interventional treatment**
- **To understand the prognoses and clinical sequela of successful lymphatic embolization of the thoracic duct and efferent lymphatics**

Chylothorax background

Chylothorax is a potentially life-threatening disease that often requires surgical intervention in dogs. Thoracic duct ligation (TDL) is most commonly performed, and can be achieved via open thoracotomy, thoracoscopy, or a trans-diaphragmatic approach.[1-4] Thoracic surgery, however, carries an inherent risk of anesthetic and surgical complications, and is often associated with postoperative discomfort or pain, requiring several days of hospitalization. Complications associated with thoracic surgery include pain, hemorrhage, infection, dehiscence, hypoxemia, and pneumothorax.[5,6] Moreover, thoracic duct ligation often requires adjunctive procedures (e.g. pericardectomy) to achieve complete resolution of chylous effusion as thoracic duct ligation will only result in clinical resolution as a stand-alone therapy in approximately 50% of cases.[1] Persistent chylothorax after ligation is thought to occur due to the formation of collateral lymphatics and the failure to ligate all branches of the thoracic duct. Thoracic duct ligation is therefore often combined with adjuvant procedures, often requiring multiple surgical approaches (e.g. laparotomy, diaphragmotomy), to achieve higher success rates.[2-8]



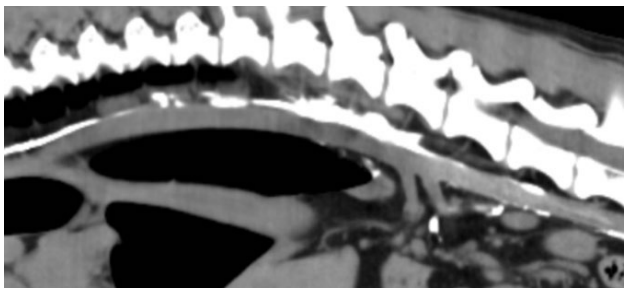
Percutaneous thoracic duct embolization is now considered superior to thoracic duct ligation alone as a method to treat chylothorax in humans as it is associated with minimal complications, high success rates, and a quick recovery period. Although non-invasive approaches to thoracic duct embolization are now well-established in human medicine, a reliable method to perform this technique has yet to be developed in veterinary medicine. Thoracic duct embolization has successfully been performed in 8 healthy dogs in an experimental study performed in 1989 (Pardo). Persistent and complete obstruction was observed in all dogs with minimal complications. There is only one report of the application of thoracic duct embolization in clinical dogs (Clendaniel). In this study, it was used as a salvage procedure for the treatment of persistent chylothorax in 2 dogs and resolved clinical signs in one of the two dogs. In the studies conducted by both Pardo and Clendanial, thoracic duct embolization was achieved via an invasive abdominal approach.

Non-invasive approaches to thoracic duct embolization have been attempted in two experimental studies. In 1996, a non-invasive, transabdominal, percutaneous approach to thoracic duct embolization with a coil was performed in one healthy dog, and no short- or long-term complications were seen (Cope). More recently, Singh et al. evaluated a percutaneous approach to thoracic duct embolization in an experimental study of 15 dogs (Singh). Due to anatomical differences with humans and the risk of inadvertent puncture of adjacent abdominal soft tissues, a lateral, percutaneous approach was deemed more appropriate for dogs in this study. Several technical difficulties were encountered with this approach, however, and, as a result, successful thoracic duct embolization was accomplished in merely 4/15 (26%) dogs.

Thus, a hybrid, laparoscopic/thoracoscopic approach to thoracic duct ligation and embolization in dogs makes sense to balance the benefits of TD ligation and embolization. The advantages are a *less invasive* technique for the surgical treatment of chylothorax in dogs, decreasing the morbidity and potential for complications that can occur with more traditional surgical approaches, including *pain, surgical site infection, hemorrhage, hypoxemia, pneumothorax and atrial fibrilation*. Additionally, thoracic duct embolization combined with ligation has not been evaluated and may lead to a *lower recurrence rate* than ligation alone by allowing occlusion of the thoracic duct over a larger area including smaller unobserved or missed thoracic duct branches.

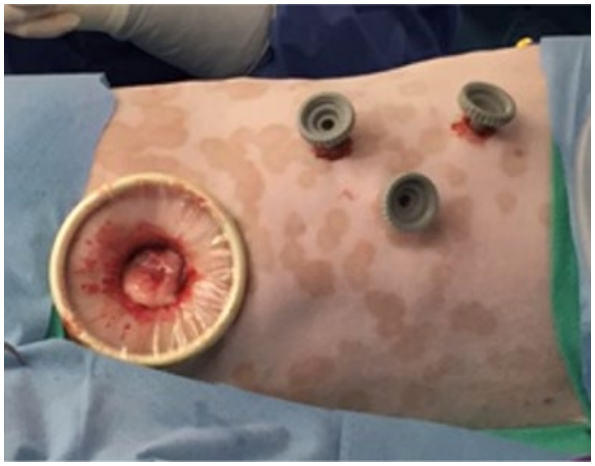
Lymphangiography

Accurate lymphangiographic assessment of the lymphatic system, cisterna chyli and thoracic duct (and branches) is critical when diagnosing and planning surgical intervention of dogs affected by idiopathic chylothorax. The current gold standard for preoperative assessment is by CT lymphangiogram (CTLa) **Fig 2**. This study is performed by CT scan following injection of aqueous, iodinated contrast (Iohexol, 350 mgI/ml; GE Healthcare Inc.) typically at 0.35 ml/kg diluted 1:1 in 0.9% saline directly into a popliteal lymph node. The patient is anesthetized and placed in sternal recumbency and the procedure is performed via ultrasound guidance or by miniature cutdown if ultrasound is unavailable. Briefly, the correct dosage of contrast is drawn up and attached to a 3-way stopcock. A 25-gauge butterfly catheter is attached to the syringe and stopcock. An assistant maintains a hold on the syringe while the radiologist injects the lymph node under ultrasound guidance. The injection is delivered slowly over 5 minutes by the assistant and CT scan performed immediately at completion. In some circumstances, a poor or incomplete study will result. This can be due to technical error, rapid clearing of the contrast from the lymphatics, or because of lymphatic abnormalities (e.g. lymphoceles or lymphatic obstruction). Other potential targets for contrast injection include: mesenteric lymph nodes, rectum, perianal tissues, diaphragm and metatarsal pad. The author has used the mesenteric lymph nodes and rectum most commonly with mixed results. In one instance, mesenteric lymph node injection resulted in enteroenteral adhesions which were documented during abdominal surgery weeks after CTLa.



Thoracic duct and efferent lymphatic embolization

Following aseptic preparation dogs are moved into the operating room and placed in sternal recumbency. The abdomen is approached via miniature laparotomy using a wound retractor through a limited, right-sided approach. An efferent mesenteric lymphatic is catheterized with a 24-gauge catheter or a mesenteric lymph node is located and a mixture of 1:1 New Methylene Blue:0.9% saline (0.05ml/kg) is injected to visualize the cisterna chyli and thoracic duct if performing thoracoscopic TD ligation or to make identification of an efferent lymphatic easier. All catheters, syringes and extension sets should be flushed with 5% dextrose in water to limit exposure of the embolic to organic elements that will cause premature polymerization. Thoracic duct and cysterna chyli embolization should be guided by fluroscopic viusualization. Most operating room C-arms are adequate for lymphangiographic assessment although newer high-resolution fluoroscopy units with digital subtraction angiography are preferred. Briefly, with fluroscopic-assistance an injection of approximately 0.10 – 0.12 mls/kg of a 1:3 mixture of n-butyl cyanoacrylate:lipiodol is performed until the caudal TD is reached (glue embolic will typically migrate craniad for a few second following pressure injection). A positive pressure breathe hold is recommended to prevent craniad migration of the embolic prior to polymerization. The injection should be continuous and delivered over 5-10 seconds and the ultimate volume will depend on the size of the dog's lymphatics. Filling beyond the mid-TD is not recommended as non-target embolization of the pulmonary vasculature may result. If TD ligation is also being performed, the TD branches identified on preoperative CTLa should all be ligated immediately following embolization. The author prefers to have the branches dissected out and isolated before embolization to facilitate rapid ligation of the TD following embolization. A final lymphangiogram should be performed to confirm immediate postoperative occlusion of the thoracic duct and visible branches.



Outcome and monitoring

If TD ligation was also performed, port site hemorrhage can happen after removal of any intercostal port. In some cases, hemorrhage from intercostal arteries has necessitated reoperation to locate and ligate the bleeding vessel. Consequently, it is strongly recommended that all port sites are monitored closely and addressed should hemorrhage be noted following port removal.

Intercostal vessel hemorrhage can result during dissection of the TD branches and can result in significant hemorrhage. It is important to have bipolar energy or vascular clips available to ligate the ruptured vessel if indicated. TD laceration is also a risk during TD dissection. The author has experienced this in a case and has been able to clip off the affected branch on either side with no adverse or on-going chyle leaks. Thoracic ducts

can be adhered to the aorta or free in the dorsal mediastinum and tend to be relatively weak and friable. Thus, caution should be exercised when manipulating and dissecting TD branches. TD embolization without ligation may make more sense in these scenarios. Short-term success for thoracoscopic TDL and pericardectomy is between 85 and 90% (Allman, Mayhew). Long-term success is not reported for a large group of dogs treated similarly although long-term success is reported to be around 70% in smaller case series. Recurrent effusion or failure for chylous effusion to resolve following surgery is most likely due to missed TD branches or formation of TD collaterals. Thus, repeat CTLa is indicated to reassess the TD anatomy in the event of surgical failure. If missed ducts or new collaterals are identified, consideration should be given to repeat embolization and/or ligation.

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