

Section 5.2.9. Small Carnivore Sampling Methods

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Objective: To safely collect biological samples from live and dead small carnivores.

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The authors assert that animal capture and sampling should always occur in compliance with all applicable laws and regulations and should only be undertaken after securing all necessary permits and approvals, including ethical approvals.

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Section 5.2.9a. Confirmation of Knowledge

When you are familiar with the information in this Guide, take the PREDICT quiz [Section 8.4.8. Small Carnivore Sampling](#).

Section 5.2.9b. Brief Overview of PPE

Minimum PPE Required for Handling Small Carnivores

The minimum PPE for sampling small carnivores includes:

- Designated clothing
- Nitrile gloves
- Protective glasses
- N95 facemask for self-protection and to avoid contaminating samples

(See the [Biosafety and PPE Guide \(Section 4.\)](#) for detailed instructions regarding PPE Use)

Note: *In order to protect both human handlers and sampled small carnivores, all personnel handling small carnivores should be vaccinated against rabies beforehand as described in the following Section.*

Section 5.2.9c. Special Considerations for Handling Small Carnivores

This section supplements [Section 5.2.5. Safe Animal Capture and Sampling](#), with which anyone handling animals is expected to already be thoroughly familiar. Note also that sampling from dead animals, whether destined for bushmeat or not, is also covered in [Section 5.2.12. Bushmeat Sampling Methods](#). However, for completeness, much of that protocol is repeated here.

Handling small carnivores involves a number of special considerations:

1. Zoonotic diseases
2. Staff vaccinations and medical concerns
3. Other hazards (bites and scratches)
4. Capture and handling

1. Zoonotic Diseases

There are numerous zoonotic diseases that may be transmitted from small carnivores to humans. Here we highlight the most important diseases that are a risk to human handlers. Research teams should be familiar with additional zoonoses that may be present in their target and bycatch species and geographic areas. It is recommended that immunocompromised people not work directly with live or dead animals.

Rabies is endemic in many carnivore populations, and may show up sporadically in any carnivore. Therefore, anyone who expects to handle live or dead carnivores should be appropriately vaccinated. Clinical signs of rabies are quite variable and can include any neurologic abnormality; this includes depressed mentation, which is a general sign displayed by nearly all sick or injured animals. When handling a carnivore, the potential for rabies should be assumed and appropriate precautions taken. Most zoonotic exposure is via bite wounds from live carnivores. However, exposure to infected bodily fluids (particularly saliva, blood and cerebrospinal fluid) must be considered when collecting samples from dead carnivores. Rabies is not transmitted by casual contact. Many other viruses can also be zoonotic.

Leptospirosis is a bacterial disease that affects a wide variety of mammals. It causes septicemia, kidney, liver, pulmonary, and/or reproductive dysfunction and is spread by contaminated water and urine. Gloves and good hygienic practices (i.e., thorough hand-washing) should provide protection.

Salmonella spp. are bacteria that can cause severe disease in humans and carnivores, but can also be carried by these species asymptotically. *Salmonella* can cause septicemia as well as diarrhea, and all carnivores are potential carriers. Special care should be taken when working with animals, live or dead, that have evidence of diarrhea. *Salmonella* spp. are most likely to be shed in the feces, but fur, traps, and vegetation where animals are held can all become contaminated. For that reason, gloves should be worn while handling animals and while cleaning/handling traps and cages in which animals have been captured or housed.

Mycobacteria bovis, the cause of bovine tuberculosis, is a bacterium sometimes found in carnivores that prey on species in which *M. bovis* is endemic. The most likely exposure for staff would be during necropsy, so gloves and mask should be worn during necropsies of carnivores in endemic areas (i.e., East Africa). This disease is of special concern for those who are immunosuppressed, but it can cause disease in immunocompetent individuals as well.

Echinococcus spp., a cestode parasite of canids and their mammalian prey, causes disease in humans when the eggs are ingested. Infective eggs are shed in the feces by the carnivore definitive hosts, which includes domestic dogs as well as wolves, foxes, jackals, lions, and sometimes other canids and felids. The disease is caused by large cysts formed by the larvae; cysts are most commonly found in the liver and lungs. For prevention, gloves should be worn and hands should be thoroughly washed after handling carnivores or items that could be contaminated with carnivore feces.

Parasites, both external and internal, may be transmitted from carnivores to humans. Internal parasites are most commonly transmitted via the feces, so precautions taken for salmonellosis should also protect against these organisms. External parasites, such as *Sarcoptes* mites (the cause of mange) and fleas, causing dermatitis, may also transmit pathogens (e.g., fleas may carry *Yersinia pestis*, the causative agent of plague). Wearing gloves, long sleeves and pants while handling carnivores can help prevent transmission of parasites. When performing

necropsies on heavily infested carcasses, the carcass may be dusted with an acaricide prior to handling.

A variety of other pathogens may be transmitted from wild carnivores to humans, and risk of exposure varies by geography and other factors. Wearing gloves, good hand washing protocols, and common sense hygienic practices will protect against transmission of most pathogens. Personnel working with animals should always wash their hands thoroughly before eating, drinking, using tobacco products, or any other activity that involves touching the face or mucus membranes.

2. Staff vaccinations and medical concerns

All those working with carnivores should be vaccinated against rabies, ensure that they have a protective titer, and be aware of appropriate post exposure prophylaxis in the case of bites.

3. Bites and scratches

With few exceptions, carnivores are predators and have formidable weapons. Small carnivores can inflict powerful bites that cause massive tissue damage and inoculate bacteria deep into tissue. Most carnivores have claws on all four feet and felids, especially, can do severe damage with their claws. Small carnivores such as civets, otters and mongooses should not be underestimated; they are remarkably agile and cannot be safely sampled without chemical restraint. No one should attempt to work with larger carnivores without proper training and a healthy respect for the risk in working with these species.

4. Capture and handling

Most carnivores are captured with traps, such as box traps, foothold traps, or snares. All traps, regardless of padding or safety features, pose some degree of hazard to the animal. Carnivores are capable of severe self-injury in any type of trap; they can break canine teeth or tear off claws in box traps, or chew off limbs caught in foothold traps. Snares set for large animals have obvious risks for smaller carnivores. For these reasons, traps should be prepared carefully, in good working order, and checked frequently, at least every 12 hours. Veterinary staff should be prepared to deal with a variety of trap injuries, including lacerations, broken teeth, and crush injuries.

Many carnivores captured in footholds or snares can be dangerous to personnel. Trapped animals should be approached carefully. Most species will need to be chemically immobilized by blowdart or pole syringe, but some smaller animals (such as some jackal species) can be handled with manual restraint and tools such as gloves and nets. Animals in box traps can be hand-injected or pole-syringed. Ketamine hydrochloride combined with a tranquilizer (such as a benzodiazepine or an alpha-2 agonist), or Tiletamine-Zolazepam (Telazol, Zoletil) are the mainstay of carnivore immobilization protocols, but many protocols have been used and protocol success varies by species. Appropriate protocols for individual species can be found in various publications such as Kreeger, Fowler, Fowler and Miller, Nielsen, and West, Heard, and Caulkett (See References Section). Chemically immobilized animals should be housed in a trap or cage until they are fully recovered from anesthesia to prevent injury or death from

drowning, falls, predation, or intraspecific aggression. While many protocols include one or more reversible drugs, neither ketamine nor the tiletamine component of Telazol are reversible, so protocols containing either of these drugs are **not** fully reversible. Therefore, giving a reversal agent is not a guarantee of an immediately awake and aware animal. Animals should be observed carefully to ensure that they are fully recovered before they are released, keeping in mind that a reactive animal is not necessarily a completely awake animal. Animals are typically ready for release when they can hold their head steady and follow movements with their eyes.

Care should be taken to decrease the stress experienced by animals prior to and during immobilization. This includes keeping your distance from the captured animal, speaking in soft voices and minimizing noise, and covering box traps with a towel or tarp to decrease visual stimuli. Similarly, anesthetized animals should have their eyes covered with a towel or cloth. It is especially critical that immediately after initial drug administration, the animal is free (to the extent possible) of visual and auditory stimuli; the quality of the induction often impacts the whole anesthetic event. During recovery, keep the caged animal within view, but at a great enough distance that the animal does not feel threatened by your presence. As always, noise should be kept to a minimum.

Anesthetized animals should be monitored regularly during recovery until they can no longer be safely handled, at which point they should be confined in a trap or cage. Essential monitoring includes measuring and recording heart rate, respiratory rate, body temperature, mucous membrane color, and pulse quality every 5-10 minutes throughout the procedure. The eyes should be lubricated with a bland ophthalmic ointment and protected from debris. Animals should be kept out of direct sunlight and overheated animals (>105°F/40.6°C) should be cooled by placing rubbing alcohol on their paws, administering cool subcutaneous fluids, or wetting their fur. Cold animals (<100°F/37.8°C) should be kept warm using tarps, blankets, or warm SQ fluids. Emergency drugs, appropriate-sized endotracheal tubes, and a mechanism for providing positive pressure ventilation (i.e., Ambu bag) should be available whenever animals are immobilized.

Section 5.2.9d. Small Carnivore Data Collection

Please refer to the three required data collection templates for data to collect. These include:

1. P2 Animal Data Collection Form
2. P2 Site Characterization Data Collection Form
3. P2 Specimen Data Collection Form

For more information on downloading templates from EIDITH see [Section 5.2.3. General Data Collection Templates and Applications.](#)

In some cases time constraints, anesthetic risk, inability to prolong immobilization, or other factors may necessitate prioritizing biological sample collection at the expense of collecting any physical measurements. At a minimum:

1. Measure and record the animal's mass (kg) initially as this can be important for proper drug dosing or emergency interventions.
2. Conduct a cursory physical exam before sampling in order to note any wounds or major abnormalities and to protect the health of both handler and animal.
3. Ensure the animal's parameters (heart and respiration rates, body temperature, etc.) are adequate for continuation of procedures. If they are not, attempts to correct them should be made and reversal of anesthesia considered if the animal's life is at risk.

Additional (Optimal) Data to Collect from Small Carnivores

The P2 data templates mentioned above are required to be filled in. Additional data and biometric measurements may be collected at the discretion of the sampling party.

Ideally, the following additional data should be collected from any small carnivores that are processed for PREDICT:

1. Body mass (weight)

Body weight: Although in an ideal world the body weight of an animal would be measured prior to drug dose calculation, because of the dangerous nature of carnivores this is rarely possible in field conditions. Weight ranges of the target (and likely non-target) species should be known before capture is attempted, and the veterinarian or biologist should have experience estimating weights. Animals should be weighed (g or kg) in bags, slings, or a suitable container using a calibrated hanging spring. (Note: If an animal exceeds the limit of spring scales, two or more scales can be linked (one hanging from the other) to distribute the weight. The total weight is the measure of both scales added together). Scales should be zeroed (checked to make sure they measure '0.0' units when empty). If scales are not available or accurate weights cannot be measured for any other reason, a weight should still be estimated but the recording sheet MUST note that it is an estimated and not a measured weight.

2. Age class (see below)

Age class: Usually the exact age will not be known. Individuals should be placed into one of the following age classes:

Age Class	Description
Juvenile	Dependent young, likely unweaned
Subadult	Animal is fully independent, appears to be sexually mature, but not fully physically mature (e.g., less than full adult size).
Adult	Animal has secondary sexual characteristics, adult size, sexually mature.
Old Adult	Adult showing signs of age degeneration (i.e. tooth wear)

3. Species Identification and Sex Determination (and reproductive status if adult female)

Species identification and sex determination: Based on morphology and unique characteristics, identify animal to genus and species (where possible) and sex. If dependent offspring are captured along with their mothers, they should be kept confined while their mother is anesthetized to prevent them from wandering off. Reintroduce them only when the mother is fully recovered.

4. Whole body photograph(s)

Photographs: At a minimum, the following digital photographs should be taken of each individual:

- a. Right and left lateral views while animal is recumbent.
- b. Full anterior facial view.
- c. Full lateral facial/head view.
- d. Views of full upper and lower dentition (which can help determine/verify age and sex).
- e. Views of any lesions (e.g., cuts, scratches), physical abnormalities (e.g., missing toes), or individually identifying marks or characteristics (e.g., healed scars, abnormal coloration, torn ears, etc.)

5. Morphometric measurements

Body measurements: Time required for collecting the biometrics (in cm/mm) should be recorded with the minimum standard mammal measurements (all linear) as follows:

- a. Head and body length (measured dorsally and linearly from tip of nose to base of tail when head is stretched and aligned with back).
- b. Tail length (from base to tip).
- c. Hind foot length (heel to tip of longest toe- exclude nail and note which toe).
- d. Tibia length ('knee to ankle').
- e. Hind foot (tarsal) length
- f. Ear length- base of the notch below the ear opening (lower rim of external auditory canal = meatus) to the most distant point of the margin of the pinna.

Additional Optional Measurements

Head length, trunk height, hip breadth, hand length and breadth, foot breadth, limb segments (thigh, lower leg, upper arm, forearm).

Chest circumference, abdominal circumference, and cranial circumference (at or above brow).

Section 5.2.9e. Small Carnivore Sampling

Capturing, trapping, darting, and immobilizing small carnivores should only be performed by experienced and skilled staff.

PREDICT personnel are expected to have detailed capture/immobilization protocols (and recording sheets, monitoring sheets, etc.) for any target small carnivore species. This sampling guide assumes a starting point of either a safely immobilized or an already dead small carnivore.

In addition to the standard PREDICT sampling and analyses, PREDICT partners are encouraged to collect additional samples and pursue routine diagnostics (e.g., blood counts and chemistries, urinalysis, etc.) where resources allow. Opportunities to collect biological samples and related health data from wild animals are uncommon and maximizing these opportunities can further advance wildlife health knowledge.

The following basic set of samples should be collected from each animal where possible (if only one sample can be collected, then place into VTM):

1. **Two oral swabs** - one in 500 μ L VTM and one in 500 μ L Trizol
2. **Two fecal samples** - one with max of 500 μ L/0.5cc feces in 500 μ L VTM and one with max of 500 μ L/0.5cc feces in 1 mL Trizol
Or
Two rectal swabs - one in 500 μ L VTM and one in 500 μ L Trizol
3. **Two whole blood samples** - one with max of 500 μ L of whole blood in 500 μ L VTM and one with max of 500 μ L of whole blood in 500 μ L Trizol
4. **Two serum samples** - 2 x 0.5 ml aliquots frozen without media
5. **Two urogenital swabs/urine samples** – one in 500 μ L VTM and one in 500 μ L Trizol

Note: *If animals are too small to collect two blood tubes (for whole blood and serum), collect serum and save remaining clot in 500 μ L VTM after serum separation.*

Freeze all samples (except tissue in formalin) in liquid nitrogen immediately in the field and transfer to -80°C freezer once back in the lab.

If there is no **short-term** access (i.e., within 24 hours) to cold chain such as in an emergency situation then samples can be collected in 500 µL of RNAlater instead of Trizol and VTM. Storage times and temperatures for samples in RNAlater are as follows:

1. 1 day at 37 °C (i.e., ambient temp)
2. 1 week in the refrigerator
3. Within one week freeze at -80 °C for storage until analysis

Sample Labeling

Tubes must be labeled with a unique specimen ID per Animal/specimen labeling guide. Please see [Section 5.2.3. General Data Collection Templates and Applications](#) for details about assigning Animal IDs.

Sample Collection from Live Small Carnivores

In most cases, live small carnivores should be chemically restrained for handling. At least two, and preferably three people are required for these manipulations: one person to monitor the animal, one to take samples, and a third to manage the tubes and record data.

- 1. Two oral swabs in VTM and Trizol** (if only one is collected, place sample in VTM): Using sterile, polyester-tipped swabs with a plastic shaft, rub the swab tip gently but thoroughly against the back of the animal's throat, saturating the swab with saliva.

Place 1 swab in a cryovial filled with 500 µL of VTM and use a flame-sterilized scissors to cut the shaft of the swab above the tip. Place the other swab into 500 µL of Trizol in another cryovial and cut shaft as above. [Note: If the plastic shaft can be snapped, then scissors are not necessary and the risk of cross-contamination is reduced. To snap the swab, lift the swab a little above the bottom of the vial then snap it. This will ensure the swab will not block the cap].

Mix each tube well. Store both cryovials in a liquid nitrogen dry shipper or dewar and transfer to -80°C freezer when possible.

- 2. Fecal samples**

500 µL or pea-sized piece of feces (200 mg) in VTM and Trizol: Collect either excreted feces, or if animal is large enough (> 1 kg) use a gloved, lubricated (saline or medical lubricant) finger to collect feces directly from rectum. Place two ~200 mg (pea size) samples of fresh feces into 2 vials, one containing 1 mL Trizol (= maximum final ratio of 1:2) and one containing 500 µL VTM (= maximum final ratio of 1:1). Homogenize by shaking. Freeze in dry shipper or dewar with liquid nitrogen and transfer to -80°C freezer when possible.

If feces are not available, collect **2 rectal swabs, one in VTM and one in Trizol:** Gently insert one sterile swab tip at a time into the animal's rectum. Rectal swabs can be moistened with

sterile saline prior to animal sampling. [Note: DO NOT USE TRIZOL AS A LUBRICANT – IT IS HIGHLY IRRITATING TO TISSUE. Also, DO NOT USE VTM AS A LUBRICANT.] Place one swab in a cryovial filled with 500 µL of VTM using a flame-sterilized scissors to cut the shaft of the swab above the tip. Place the other swab into a tube with 500 µL of Trizol. Store in a dewar or dry shipper with liquid nitrogen dry shipper and transfer to -80°C freezer when possible.

3. Whole blood and serum samples

Precautions

- **At least one person present should have previous experience in small carnivore venipuncture to avoid injury to the animal.**
- Animals should be immobilized using either injectable or gas anesthesia according to appropriate guidelines. On occasion some species may be manually restrained for venipuncture but extra care must be taken to avoid injuries to personnel and animals.
- The person restraining the animal is responsible for monitoring respiration and other vital signs and communicating the status of the animal appropriately.
- No more than 1 ml of blood per 100 g (= 10 ml/kg or 1%) of body weight should be collected at any one time; it is best to limit collection to 0.6 ml blood per 100g.
- Blood should always be considered highly infectious and hazardous.

Collection Procedure

1. Select appropriate venipuncture site:

Animal family	Venipuncture site
<i>Felids</i>	<p>Medial saphenous vein: With compression of the inner thigh, this vein can be prominent and superficial, but often collapses during collection. Use of a butterfly needle and extension set may help avoid this problem, as well as using a smaller syringe and pulling back slowly on the plunger.</p> <p>Cephalic vein: In larger species the cephalic vein might be large enough for safe blood collection.</p> <p>Jugular vein: This may be the only option in very small animals and must be accessed carefully.</p> <p>Lateral tail vein: This may be accessed in larger felids; the same comments as for the medial saphenous vein apply.</p>

Canids and Hyenids

Lateral saphenous vein

Jugular vein

Cephalic vein

Femoral vein: It is easy to hit the femoral artery instead; if this happens,

be sure to apply firm, direct pressure for several minutes to effect hemostasis

Medial saphenous vein: May be accessible in some species

Mustelids

Jugular vein: Due to their short muscular necks, this can be a difficult vein to access. Placing pressure in the thoracic inlet bilaterally often helps to occlude this vein and help it pop up.

Femoral/saphenous vein: These tend to be short and difficult to hit, and the femoral artery can be accidentally hit as in canids and hyenids.

Viverrids and Herpestids

Jugular vein: Best option for adequate samples

Cephalic: Small volumes

Tail (ventral midline): Small volumes

Ursids

Cephalic vein

Saphenous vein

Jugular vein

Note: When using an alpha-2 agonist in a chemical immobilization protocol, the peripheral veins often collapse due to vasoconstriction. Using the jugular vein will often be necessary for venipuncture.

2. Select appropriate sized needle and syringe (or vacutainer) for the size of the animal.
3. Disinfect the site with iodine solution or alcohol.
4. Collect sample.
5. **Do not recap needle.**
6. Apply pressure to site of bleeding using a cotton ball or gauze pad until bleeding ceases (approximately 1 minute).
7. Process blood (see below).
8. Properly dispose of sharps and other biohazard materials immediately upon transfer of sample to collection vials.

Blood Processing

- a. **Whole blood in EDTA:** Collect 1 tube of whole blood in EDTA (lavender top vacutainer). Add up to 500 µL of whole blood into 2 vials, one containing 500 µL Trizol and one containing 500 µL VTM (= maximum final ratio of 1:1) and mix each vial well. Place vials into liquid nitrogen in dry shipper or dewar and transfer to -80°C freezer when possible.
- b. **Aliquot serum into cryovials:** Collect blood into a serum tube (red top or tiger top, if >1 mL blood is collected, or into 1.5 mL conical Eppendorf tubes). Place labeled blood tubes

in a rack on ice (optimally) for up to 2 hours prior to centrifuging. Centrifuge the blood samples. If a centrifuge is not available, red top tubes with blood can be left standing on ice overnight to allow serum to separate. Use a pipette to draw off serum, aliquot into 0.5 mL volumes per cryovial, and store cryovials in a dry shipper or dewar. As soon as possible, remove samples and place in cryoboxes and store in an -80 °C freezer.

4. Urine

Collect 2 urogenital swabs and place one in 500 µL of VTM and one in 500 µL of Trizol. Swabs can be moistened with sterile saline prior to animal sampling. Alternatively, if the animal urinates, urine may be collected by free catch from the urethra. Aliquot up to 500 µL of urine using a pipettor into one cryotube and add 500 µL of VTM. Add another sample of up to 500 µL of urine into another cryotube with 500 µL of Trizol. Mix cryotubes well.

Sample Collection from Dead or Euthanized Small Carnivores

If carcasses are not whole, then the [Bushmeat Sampling Methods \(Section 5.2.12.\)](#) may be more applicable. If bodies are relatively whole and fairly fresh, then sample as described above. If an animal must be euthanized due to humane or veterinary care reasons, see [American Veterinary Medical Association guidelines \(Section 8.5.2.\)](#).

As discussed throughout this protocol, all wildlife should be considered potentially infectious for a wide variety of dangerous pathogens and dead animals in particular should be sampled only following all safety measures including proper PPE use, proper work station decontamination, and proper carcass disposal as outlined here and in other PREDICT documents.

Though not required for PREDICT sampling, thorough necropsy procedures can be very beneficial and might pertain to some animals (e.g., valuable or known individuals, suspicious deaths, etc.). Necropsy protocols are addressed in separate documents. Time and skill permitting, when full necropsies are performed, following any Association of Zoos and Aquariums/AZA (or similar) necropsy protocol is recommended and most can be adjusted for application to other species. (*Note that properly following extensive necropsy procedures and collecting and measuring all samples can require 4-6 hours for a single animal.*)

Post-Mortem Blood Collection

From recently dead animals, it may be possible to collect whole blood (often clotted) from the right side of the heart where the largest volume of blood is available. Collect all available blood into an appropriate size container (typically one or more blood tubes). Allow the tubes to sit undisturbed for at least 30 minutes, and then centrifuge at high speed (2000 x G for 20 minutes). Transfer the serum (clear, yellow or red-tinged fluid at the top), preferably via pipetting, to appropriately labeled cryovials. Transfer the remaining blood clots to separate cryovials. Refrigerate or freeze both the serum and blood clots.

If a centrifuge is not available, allow the clots and cells to settle as much as possible, and then collect the serum and clots as described above. If the animal's death is recent enough that the blood has not yet clotted and a centrifuge is not available, invert the blood tubes after the blood has been collected to allow the clot to form on the rubber stopper. After the blood has clotted, turn the tube right side up and carefully remove the stopper with the adhered clot, thereby leaving a clean serum sample in the tube.

At a minimum, as many of the following blood samples as possible should be collected:

- 2 samples of 500 μ L (**whole blood**) placed in 2 vials, one containing 500 μ L **Trizol** and one containing 500 μ L **VTM** (= maximum final ratio of 1:1). Mix each vial well. **If only one sample can be collected, then place it into VTM.**
- 2 or more aliquots (0.5 ml) of **separated serum**, frozen

Tissue Collection

Collect **three**, adjacent, approximately 200mg (pea-sized) samples of the following tissues:

- Adrenal
- Colon
- Heart
- Liver
- Lymph node
- Ovary
- Testes
- Cecum
- Duodenum
- Kidney
- Lung
- Spleen
- Pancreas
- Other, if required*

One specimen should be frozen in 500 μ L VTM in a cryovial, one should be frozen in 1 mL Trizol in a cryovial, and one should be stored at room temperature in a small vial or jar in 10% buffered formalin at a volume of fixative 10 times the volume of the tissue (once fixed, the tissue may be transferred to a smaller volume for shipment). **If only one sample can be collected, then place it into VTM.**

*It will usually require experience to identify abnormal tissues, but potentially recognizable gross lesions include masses, discolored areas, ulcerations, etc. Samples for histopathology (i.e., in formalin) should be collected at the abnormal margins to include both normal and abnormal sections in the same piece of tissue. Collection of any obvious internal parasites in ethanol is also recommended.

Section 5.2.9f. References

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