Blood Collection Techniques in Exotic Small Mammals

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Abstract

Blood collection from small exotic pocket pets can be difficult to achieve. The individual collecting the blood must know both the anatomy and behavior of the species to obtain suitable amounts of blood for diagnostic testing. Given the animals' small size, it is often difficult to collect large volumes of blood. A clinician serious in developing an exotic small mammal practice should understand the limitations of blood sample collection and the risks involved with the procedure. Unlike domestic animals, these pets are often not comfortable with being handled and are often prone to induced complications when presented to a veterinary clinic and restrained for examination. For some cases, the clinician will have to determine if the risk of getting the sample is better achieved by anesthetizing the patient, and if doing so will have a detrimental effect on the animal. One will also need to consider the effect of the anesthetic versus the stress the restraint may have on the blood results. Copyright 2009 Elsevier Inc. All rights reserved.

Key words: blood collection; exotic small mammal; ferret; rabbit; rodent; venipuncture

The size of many small exotic pocket pets seen in private veterinary practices can make diagnostic blood sample collection problematic. It is often difficult to access veins or arteries of adequate size to collect sufficient blood for diagnostic testing. However, with the advent of in-house analyzers that can measure hematologic and blood chemistry parameters from small volumes of whole blood (50-100 µL; 0.05-0.1 mL), it is now possible to pursue diagnostic blood work on many of these exotic small mammals.

Minimizing the Effects of Stress and Other Effects on Blood Results

The majority of the small exotic mammals that present to veterinary clinics are prey species by nature, with the ferret (Mustela putorius furo) being an exception. Therefore, these animals are easily stressed when handled, anesthetized, or transported. Furthermore, while at the veterinary clinic, these animals are often exposed to bright lights and loud noises and can hear, smell, and see predators such as dogs and cats which are disturbing to these often nocturnal and crepuscular species. If one can minimize or eliminate the effect that outside stressors have on these animals, they can reduce these effects on stress-sensitive blood parameters.

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Understanding the animal’s natural behavior and making appropriate accommodations is also helpful when collecting blood samples from exotic small mammals. For species such as sugar gliders (Petaurus breviceps), which are mainly nocturnal, it is preferable to schedule the examinations early in the morning when they are less active and when the clinic is quiet. However, if you want to observe the same animal’s activity levels, the appointment should be scheduled in the early evening when they are more active. On arrival at the clinic, these animals should be brought directly into a warm (70-72°F, 21-22°C) examination room with subdued lighting, away from other disturbing sights, sounds, and smells that are associated with veterinary hospitals.

For those small exotic mammals that are used to being handled, blood collection can be done quickly with minimal restraint. Ideally, the owner should transport the animal to the clinic in its own cage, covered with a towel so that it is in a darkened enclosure. The animal should be removed from its cage for venipuncture and then returned to its cage rather than being put into an unfamiliar enclosure. For animals that are not easily restrained, anesthesia is recommended to facilitate sample collection. Traditionally, small exotic, pocket pets have been bled using manual restraint but the author believes that in the majority of the cases anesthesia is indicated in these animals. The author prefers to anesthetize the animal in its cage before handling and then return it directly to its cage to recover.

Conditioning an animal before sample collection can reduce the potential effects of stress on the results. A study by Fluttert and coworkers demonstrated that rats handled for 2 minutes for 4 to 5 days before blood collection produced levels of plasma corticosterone 8 to 10 times lower than those in rats not preconditioned. Furthermore, corticosterone levels in the nonpreconditioned rats did not return to normal until 120 minutes later. The preconditioning of the rats consisted of placing the rat in a towel, loosely folded around the animal, for 2 minutes, thereby allowing the subject to explore the “tunnel.” The rat’s tail was stroked and gently squeezed to simulate the blood collection. After the preconditioning period, the blood sample was collected. This type of preconditioning can easily be done by the pet’s owner before it is brought to the clinic. The towel used to handle the animal during preconditioning can be used for the blood collection, because it will have the animal’s scent on it and should minimize the stress level of the patient. If a syringe case with holes in the end is used for restraint, the owner should be instructed to precondition the rat for the procedure by putting treats in a similar syringe case.

There is evidence that the stress of anesthetic induction can have an effect on blood values in laboratory animals. For example, ferrets anesthetized with isoflurane (Forane; Baxter Health Care Corporation, Deerfield, IL USA) exhibit a rapid decrease in their hematocrit, hemoglobin, and red blood cell count, and these hematologic values do not return to preanesthetic levels until 45 minutes after the initiation of the procedure. The clinician must therefore consider the effect that anesthesia has on laboratory data and the risk of the anesthesia on an ill ferret. It is ultimately the veterinarian’s responsibility to determine the benefits and risks of using anesthesia for these animals when collecting blood for diagnostic testing.

Many of the blood collection sites and sampling techniques used for small exotic mammals are similar to those described for cats and dogs. However, for some of the blood collection sites, such as the cranial vena cava, an inexperienced handler and phlebotomist would benefit from anesthetizing the patient until the necessary skills are acquired to perform the procedure on an alert animal. Doing so reduces the stress on the animal, the owner, and the veterinary personnel performing the procedure.

There are a number of other physiologic and environmental factors that can affect hematologic test results, including gender, age, strain, circadian rhythms, stage of reproductive cycle, pregnancy, diet, and season (e.g., animals that hibernate such as a hamster). Laboratory processing (e.g., type of anticoagulant used) and venipuncture site can also affect the blood test results. It is important for clinicians to consider the potential effects of these factors when interpreting test results. Ideally, if one’s practice has a large enough small exotic mammal caseload, then in-house reference ranges can be developed, but this requires one to follow a consistent methodology when collecting blood samples including the use of anesthesia, type of anesthetic agent, and form of anticoagulant.

**Basic Restraint and Anesthesia for Blood Collection**

Basic handling and anesthetic protocols for small exotic mammals have been well covered in several recent articles and books, and thus will not be covered in this article. Specific restraint procedures for handling of small exotic mammals for collecting blood from sites unique to these patients will be described.
Total Blood Volume and Recommended Maximum Blood Sample Size

The total volume of blood that can be safely collected from small exotic mammals for blood analysis should be based on the size of the animal being bled. A standard of collecting 1% of the animal’s lean body weight is often given as the best estimate for a safe sample volume; however, there are a number of cases where sampling at this volume is not recommended. For example, it is generally recommended that smaller volumes of blood be collected for patients that are geriatric or suspected of being anemic or hypoproteinemic. It is important for the veterinarian to consider the potential negative effects of collecting too much blood from a compromised or obese patient.

Ideally, the amount of blood collected from a patient should be based on published data of the total blood volume of a given species. However, before using the published figures, one needs to know the methodology used in determining this data. Depending on the methodology used, published calculations of total blood volume can vary up to 10%.

The most accurate techniques use sodium radiochromate or sodium perethionate as a red cell label to determine red cell volume, and human serum labeled with radioiodine as a plasma label to measure plasma volume.14 Table 1 gives published figures on whole blood volumes and whole blood volume as percentage of body weight; unfortunately, the methodology used to calculate these data is not described.7,14,15

There are other physiologic factors that can affect the volume of blood that can be safely collected from a patient. For example, as certain species of animals increase in size, their percentage of blood volume decreases (Table 2); therefore, a range of percentages is required for accurate interpretation.14 In addition, there can be differences in total blood volume when comparing different species and age of the animal. The most accurate estimates of blood volume are made based on lean body mass or surface area, but performing these calculations in a busy veterinary hospital is impractical.14 It is best for the veterinarian to err on the side of caution by using a lower blood sampling volume estimate (0.8%) of lean body mass and collecting that volume no more than once every 14 days. Table 3 compares the sample volume that can be collected when limited to 0.08%, 1%, or 10% of the animal’s body weight for some small exotic mammals.16

Blood Collection Procedures

Although it can be difficult to collect blood from the small veins of exotic mammals, one can take some consolation in knowing that there are similarities in the vascular anatomy between these animals and larger domestic species that allow for similar approaches for collecting blood samples.16 Some of the difficulties encountered with exotic mammal veni-
puncture can be attributed to anatomy, such as a short neck or tail. In many cases, success with sample collection can be achieved by changing one’s approach to collecting the sample. For example, instead of targeting the jugular vein as it courses along the neck, one can collect the blood from the site where the jugular divides on the animal’s cheek. There are a number of good resources for identifying the information available to the veterinary practitioner. The Blood Sampling Microsite by the National Centre for the Replacement, Refinement and Reduction of Animals in Research provides a review of blood collection techniques for the rat, mouse, guinea pig, hamster, ferret, and rabbit. A link for that site can be found at: http://www.nc3rs.org.uk/bloodsamplingmicrosite/page.asp?id=313.

Animals that are not used to being handled, or which are otherwise difficult to restrain, should be anesthetized for physical examination and blood collection. For any species that the veterinarian is unfamiliar with, isoflurane anesthesia should be considered. However, isoflurane can adversely affect the results of blood parameters in some exotic small mammals (e.g., ferrets), and these limitations must be considered when interpreting the results. Blood collection on an alert animal can avoid the potential effects that anesthetic agents have on the laboratory test results.

Ideally, animals should be fasted for several hours before collecting blood, in the event anesthesia is needed for blood collection or collection of samples for other diagnostic procedures and also to minimize the effect that a recent meal may have on test results. One must also consider the medical problems that the animal presents with and risks that fasting may have on the animal’s health.

To maximize the recovery of a sample, blood can be collected from the hub of the needle. Before collecting a blood sample, heparin can be drawn up in the needle and the excess expelled from the hub of the needle. Because anticoagulants can affect red blood cell morphology, it is also important that some blood smears be prepared without anticoagulants at the time of blood collection.

Laboratory results may vary depending on whether serum or plasma is submitted for analysis. One should always verify with the diagnostic laboratory which sample type will yield the most valid test result. The plasma to blood cell ratio should be estimated at 1:1, but the yield of plasma from a blood collection may only be up to one third of the total sample.

### Table 3. Body weight, whole blood volume, sample volume limited to 0.08%, 1% or 10% of body weight of small mammals

<table>
<thead>
<tr>
<th>Species</th>
<th>Body Weight (g) M/F</th>
<th>Total Blood Volume (mL)</th>
<th>Sample Blood Volume Limited to 0.8% Body Weight (mL)</th>
<th>Sample Blood Volume Limited to 1% Body Weight (mL)</th>
<th>Sample Blood Volume Limited to 10% Blood Volume (mL)</th>
</tr>
</thead>
<tbody>
<tr>
<td>Mouse—male</td>
<td>20-40</td>
<td>1.6-3.2</td>
<td>0.2-0.3</td>
<td>0.2-0.4</td>
<td>0.16-0.3</td>
</tr>
<tr>
<td>Mouse—female</td>
<td>25-63</td>
<td>2.0-2.6</td>
<td>0.2-0.5</td>
<td>0.3-0.6</td>
<td></td>
</tr>
<tr>
<td>Rat—male</td>
<td>267-520</td>
<td>20-40</td>
<td>2.1-4.2</td>
<td>2.7-5.2</td>
<td>0.2-0.4</td>
</tr>
<tr>
<td>Rat—female</td>
<td>250-325</td>
<td></td>
<td>2.0-2.6</td>
<td>2.5-3.3</td>
<td></td>
</tr>
<tr>
<td>Hamster—male</td>
<td>85-130</td>
<td>6.8-12</td>
<td>0.7-1.0</td>
<td>0.85-1.3</td>
<td>0.7-1.2</td>
</tr>
<tr>
<td>Hamster—female</td>
<td>95-150</td>
<td></td>
<td>0.8-1.2</td>
<td>0.95-1.5</td>
<td></td>
</tr>
<tr>
<td>Gerbil—male</td>
<td>45-130</td>
<td>4.4-8.0</td>
<td>0.4-1.0</td>
<td>0.45-1.3</td>
<td>0.4-0.8</td>
</tr>
<tr>
<td>Gerbil—female</td>
<td>50-85</td>
<td></td>
<td>0.4-0.7</td>
<td>0.5-0.9</td>
<td></td>
</tr>
<tr>
<td>Guinea pig—male</td>
<td>900-1200</td>
<td>40-80</td>
<td>7.2-9.6</td>
<td>9-12</td>
<td>4-8</td>
</tr>
<tr>
<td>Guinea pig—female</td>
<td>700-900</td>
<td></td>
<td>5.6-7.2</td>
<td>7-9</td>
<td></td>
</tr>
<tr>
<td>Chinchilla—male</td>
<td>400-600</td>
<td>70</td>
<td>3.2-4.8</td>
<td>4-6</td>
<td>7.0</td>
</tr>
<tr>
<td>Chinchilla—female</td>
<td>450-800</td>
<td></td>
<td>3.6-6.4</td>
<td>4.5-8.0</td>
<td></td>
</tr>
</tbody>
</table>
collected depending on the physiologic state of the patient.\textsuperscript{18}

In small exotic mammals, restoration of blood volume after sample collection usually occurs within 24 hours; however, it could take longer depending on the life span of the red blood cells being examined. In some cases it may take up to 2 weeks or longer for the hemoglobin, hematocrit, and total red blood cell count to return to normal. If blood loss exceeds 20\% to 25\% of total blood volume, it can lead to hypovolemic shock and death if not treated immediately. It is, therefore, imperative that hemostasis after blood collection is confirmed and that staff are familiar with the signs associated with stress, anemia, and shock so that immediate treatment can take place if any of the critical signs are noted (Table 4).\textsuperscript{19}

Many of the smaller exotic mammals have veins too small to bleed with a needle and syringe. It may be necessary to punch or lance the blood vessel, or, if the vein cannot be seen or felt, it may need to be accessed using certain landmarks and doing a “blind stick.” The skin over the site should be aseptically prepared by cleaning with a chlorhexidine gluconate solution or scrub (ChlorhexiDerm Disinfectant Solution or ChlorhexiDerm Plus Scrub; DVM Pharmaceuticals, Inc., Miami, FL USA) or 70\% alcohol (isopropyl alcohol 70\%; Veterinary Products Laboratories, Phoenix, AZ USA) followed by the application of a topical anesthetic cream (lidocaine 2.5\% and prilocaine 2.5\%, EMLA cream; AstraZeneca LP, Wilmington, DE USA) over the vein 30 minutes before the blood collection procedure.

RestRAINT tubes can be used for several of the small exotic mammals, and it is important to wash and disinfect these devices between sessions to remove pheromones and to minimize the spread of disease. If the animal that is being restrained has an infection or is stressed by being in the tube, the patient may release pheromones, which can affect the next animal placed in the restraint device. Only an appropriate-sized tube for the individual animal should be used. Monitoring of the patient is necessary to prevent hyperthermia or the development of respiratory compromise due to malposition within the restraint tube.

\textbf{Venipuncture Techniques in Exotic Small Mammals}

\textbf{Mice (\textit{Mus musculus})}

To circumvent their small size and short neck, a number of anatomic sites have been identified to collect blood samples in mice.\textsuperscript{20} Most of these sites have been developed in research settings where many mice are bled one right after another. With training, staff can quickly become proficient in accomplishing difficult blood-collecting procedures and minimize the stress on the mouse. Some of these collection sites may not be routinely suited for veterinary hospitals but are included here as a possible technique that can be used in an emergency.

It is highly recommended that isoflurane anesthesia be used for blood collection for the mouse patient, a species very prone to being stressed by transport, restraint, and being moved into a new cage. In one study, transportation caused increased corticosterone levels in mice for 48 hours,\textsuperscript{1} whereas in another study, the corticosterone levels rose after a brief 12-minute transport and the white blood cell count significantly decreased at 4 hours and returned to normal only 12 hours after transport.\textsuperscript{21} If there is a problem obtaining blood from one site, other sites may then be quickly accessed without prolonging physical restraint and thus further stressing the mouse and affecting the laboratory test results. If possible, the mouse should be anesthetized in its home cage and allowed to recover in the same cage. By consistently using anesthesia for blood collection, the effects that the anesthetic has on the blood test results will be “standardized” across all of the individual mice sampled, rendering in-house blood reference ranges consistent for comparison.

\textbf{Venipuncture Sites Recommended for Private Practice}

\textit{Lateral Saphenous Vein.} Increasing a mouse’s body temperature will help to dilate blood vessels before bleeding.\textsuperscript{22} Warming the mouse can be accomplished by placing the animal’s cage 6 to 8 inches under a low-watt (100-W) light bulb, placing the cage on a heating pad set at a low setting, or placing the cage in an incubator at 102°F (39°C) for 5 to 10 minutes. The mouse should be observed frequently while being warmed because overheating will affect blood test results and can lead to dehydration and hyperthermia. The incubator should be calibrated frequently and monitored for any hot spots.

A mouse can either be anesthetized with isoflurane or manually restrained for blood collection. If general anesthesia is not used, a topical anesthetic cream (e.g., EMLA cream) should be applied over the vein 30 minutes before blood sampling. Commercially available restraint tubes are produced for research and can also be used to restrict the movement of an animal for venipuncture. A source for companies supplying restraint tubes is the Labora-
Other alternatives for mouse restraint devices are an appropriate-sized plastic centrifuge tube or syringe case (35 mL) that has 5 holes punched or drilled in the end for ventilation. The tube should be rendered opaque or covered in the area of the animal’s head to calm the animal. Masking tape is normally placed over the open end to partially cover the tube and allow the rear leg to be pulled out while keeping the animal in place. To prevent the sticky side of the tape from contacting the mouse’s fur on the inside of the tube,

| Table 4. Species-typical signs of pain and distress in laboratory animals¹⁹ |
|---------------------------------|-----------------|---------------------------------|
| **Species** | **Mild to Moderate Pain/Distress** (Use Butorphanol, Buprenorphine, or Ibuprofen) | **Severe or Chronic Pain/Distress** (Use Morphine or Oxymorphone) |
| Mouse | Eyelids partially closed; rapid, shallow respiration, grunting or chattering on expiration; decreased grooming; piloerection; increased vibrissal movements; unusually apprehensive or aggressive (tend to bite); cannibalize neonates; possible writhing, scratching, biting, self-mutilation; hunched posture; decreased movement; ‘waddle’ gait; sudden running movements (escape); aggressive vocalization (may be ultrasonic) when handled or palpated; guarding; decreased activity | Weight loss; dehydration; incontinence; decreased grooming; soiled hair coat; eyes sunken, lids closed; “slobbers”; wasting of muscles on back; sunken or distended abdomen; abdominal writhing; anorexic; decreased vibrissal movements; unresponsive; separates from group; hunched posture; shortened gait; sleeping on its side; ataxia, circling; hypothermia; decreased vocalization |
| Rat | Eyelids partially closed; porphyrin staining around eyes, nose; piloerection ± hair loss; increased aggression; reduced exploratory behavior; aggressive vocalization (squeals) when handled; licking, self-mutation (biting and/or scratching) guarding; decreased activity | Eyes closed; dehydration; anorexic; weight loss; incontinence; soiled hair coat; depressed/unresponsive; sunken or distended abdomen; self-mutilation; decreased vocalization; hypothermia; “slobbers” |
| Syrian Hamster | Ocular discharge; increased aggression; hunched posture; reluctance to move | Hair loss; weight loss; lethargic change in sleeping patterns; lateral recumbency; hypothermia; sores on lips, paws |
| Gerbil | Ocular discharge; may “faint” when handled; changes in activity and burrowing behavior; arched back; hunched posture; aggressive (tend to bite) | Weight loss; facial sores; hair loss on tail |
| Guinea pig | Eyes sunken and dull; changes in respiration; quiet; inactive; lethargic; hunched posture; increased vocalization when handled | Weight loss; hair loss; dehydration; unresponsive; increased barbering; loss of righting reflex; decreased vocalization; hypothermia |
| Rabbit | Apprehensive; anxious; hide or face the back of the cage; aggressive; scratching; licking; white discharge from eyes, nose; wet fur on inside of forelegs; drags legs; increased respiratory rate; piercing squeal; cries; cannibalize neonates | Repetitive vocalization (squeal, cry); teeth grinding (bruxism); anorexic; hunched; reluctant to move; piloerection; labored breathing; weight loss; increased salivation; winching; abdominal contractions |
| Ferret | Reluctance to sleep curled up, sleeps laying out on its side; tucked abdomen; stiff gait; increased respirations; holds head elevated or down and extended forward; lethargic; anorexic; teeth grinding | Reluctant to move; eyes partially closed; dull eyes |

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an additional square piece of masking tape should be used to cover the adhesive at this location.

For optimum exposure of the vein, the fur on the caudal surface of the mouse’s thigh is clipped and the skin cleaned with alcohol. Petroleum jelly (Vaseline; Unilever United States, Inc., Englewood Cliffs, NJ USA) or silicone grease (McNett Silicone Grease 100% Pure Silicone Lubricant; McNett Corp., Bellingham, WA USA) is then applied over the vein to prevent the blood from spreading away from the site into the fur and thus aid in pooling the blood for easier collection. A tourniquet (e.g., rubber band clamped with a hemostat) can be placed just proximal to the stifle, and gentle pressure and tension can be applied at the same site to stretch the skin over the hock and stabilize and dilate the vein. Instead of using a tourniquet, the fold of skin between the tail and thigh can be grasped between the thumb and the index finger to extend the leg and hold off the saphenous vein.

The lateral saphenous vein can be entered or pricked with a 22- to 30-gauge needle and the pooled blood collected with a microhematocrit tube (BD Plastic Clad Microhematocrit Tubes; Becton, Dickinson and Company, Franklin Lakes, NJ USA). To up to 0.2 to 0.3 mL of blood can be safely collected from mice. The foot and leg are then flexed and/or dry gauze is used to supply slight pressure to aid in hemostasis. Later, additional samples can be collected by removing the blood clot or scab from the area. Pictures of the procedure can be found on the Vivarium of the University of Bergen website at: http://www.uib.no/dyreavd/Vivarium-blood-sampling.pdf.

Submandibular Veins. Slightly behind the mandibular joint, the orbital and submandibular veins join to form the jugular vein. Where these vessels meet is a site from which one can collect 0.2 to 0.5 mL of blood using either general anesthesia or manual restraint (Fig 1). If manual restraint is performed, the mouse should be scruffed with the thumb and index finger to stabilize the submandibular vein while the little finger holds the base of the tail to the palm of the hand. An 18- to 23-gauge needle, #11 scalpel blade, or a mouse-bleeding lancet (Goldenrod Animal Lancet; MEDIpoint Inc., Mineola, NY USA) can be used to puncture the skin at a 90° angle to the skin. The lancet, needle, or blade must be quickly removed to collect the maximum amount of blood. Blood can be collected with either a microhematocrit or Microtainer tube (BD Microtainer Blood Collection Tubes; Becton, Dickinson and Company). Once the blood sample is collected, the tension on the skin of the neck can be relaxed slightly and light pressure with dry gauze can be applied to the punctured area for hemostasis. Several samples can be collected in one day using this method by removing the scab or blood clot or lancing the vessels on the other cheek. The potential risks associated with collecting samples with this method include inadequate penetration (i.e., too shallow or too deep), lacerating the skin and causing tissue damage, compromising the buccal surface and thereby causing bleeding into the mouth, or lancing too high above the vein resulting in bleeding into the ear canal. When using a needle to lacerate the vessel wall, it is possible for the blood to coagulate in the needle, leading to a smaller blood sample. The Goldenrod Animal Lancet addresses all of these potential problems by allowing for control of the depth of the puncture, thereby preventing too superficial or too deep penetration into the tissue. Information on the lancet, directions, and a video showing the procedure can be found at: http://www.medipoint.com/html/directions_for_use2.html. The commercial lancet is available in several sizes, and selection should be based on the size and age of the mouse (e.g., 4.0 mm for 2- to 6-week-old mice, 5.0 mm for 2- to 9-month-old mice, 5.5 mm for 9-month and older mice). Information regarding commonly asked questions about the use of the lancet for mice can be found at: http://www.medipoint.com/html/whatsizefrequent_questions.html.

Femoral Vein, Medial Saphenous Vein. To collect blood from the femoral or medial saphenous vein, it is preferable that the mouse be anesthetized or, if not anesthetized, restrained in a tube as previously described for the saphenous vein. If only one person is
going to restrain the animal and collect the sample, then the mouse should be grasped by the skin behind the shoulder blades with the thumb and index finger of the nondominant hand, the body of the mouse held with its dorsum against the palm of the hand, the skin of the mouse’s upper thigh held between the second and third fingers to block venous return, and the last finger used to hold down the tail against the palm of the hand. A topical anesthetic cream (e.g., EMLA cream) should be applied to the area being lanced 30 minutes before the procedure. The fur should be shaved over the femoral and/or medial saphenous vein and the area disinfected with alcohol, after which petroleum jelly or silicone grease should be applied over the vein. A 25- to 26-gauge needle without a syringe attached should be bent to a 30° angle at the hub and placed into the vein. Blood is collected with a microhematocrit tube directly from the needle hub or from a location at the proximal part of the vein that has been punctured at a 45° to 90° angle. The femoral vein lies parallel and caudal to the femoral artery and nerve; therefore, care must be taken to avoid these structures. If one cannot visualize the vein, then its location can be determined by palpat ing the femoral pulse and looking for the vein lying next to the artery. Dry gauze should be applied to the vein puncture site for hemostasis, and the animal should be monitored for 5 to 10 minutes after the procedure. Additional samples can be taken by removing the blood clot or scab.

When anesthesia or a restraint tube is used, the animal’s leg can be pulled away from the body wall, exposing the medial thigh. This allows the phlebotomist to place a cotton swab in the inguinal area and apply pressure to hold off the vein. The rest of the procedure is as described above.

**Jugular Vein.** The jugular vein can be used to collect blood from an adult mouse, but success may be limited because of the animal’s size. For jugular venipuncture, the mouse should be anesthetized with isoflurane and held in the palm of the nondominant hand. A loop of string from a dry gauze square can be looped around the upper incisors so that the head can be pulled dorsally with the gauze. The ventral surface of the animal’s neck should be shaved and the site disinfected and dampened with alcohol. The jugular veins will appear as blue or deep blue lines running from 2 to 4 mm lateral to the junction of the sternum and the clavicle up to the angle of the jaw. Blood is collected with a 25-gauge needle attached to a 1-mL syringe or a 25-gauge needle alone. It helps to bend the needle at the hub about 30° to better access the vessel. If the jugular vein cannot be visualized, insert the needle 2 to 4 mm lateral to the sternoclavicular junction at a depth of 1 to 3 mm in the direction of the angle of the jaw. Stabilization is achieved by passing the needle through muscle. Up to 0.2 to 0.3 mL of blood can be collected safely from a mouse jugular vein. Gentle pressure with dry gauze should be applied over the venipuncture site for hemostasis.

The anesthetized mouse can also be placed on a sloped, rectangular restraint bleeding board. The board is folded in the middle on the long axis, which creates an angle of 30°. At each end of the fold are 2 pieces of string used to tie the mouse’s forelimbs. The anesthetized mouse should be placed in dorsal recumbency with its back lying parallel to the long axis of the board, and with its neck lying on the fold so that its head can be extended dorsally. The mouse’s front legs should be secured with the string and tied out laterally from the body. The body and the rear legs should be supported by an assistant so the mouse does not slip down the sloped board. The animal’s breathing should be monitored. The fur should be clipped over the neck and the site prepared aseptically. The procedure for collecting the sample from the jugular is the same as described above.

**Lateral Tail Vein.** The mouse should be anesthetized for the procedure and placed in lateral recumbency. The lateral tail veins are observed on the lateral aspect of the tail at its base (Fig 2). If it is difficult to
see the veins, illumination may be used to assist with visualization. The veins will need to be dilated with the techniques described for the saphenous vein or by placing the tail in warm water (95-104°F, 35-40°C) for 5 to 10 seconds. The disadvantage of using the tail vein is that once entered, it stimulates the sympathetic nervous system, causing vasoconstriction of the vessel.25

To obtain access to the tail vein, the vein should be held off at the base of the tail. This can be accomplished with a tourniquet designed by Mina-sian, which can be made from a 2-, 5-, or 10-mL syringe and 20 to 30 cm of 2-0, 3-0, or 4-0 silk, linen, or nylon suture.26 To make the tourniquet, remove the plunger from the syringe and pass a needle and thread through the injection port of the syringe, leaving 1 to 2 inches of the thread hanging out. The needle and thread are then passed through the top of the barrel and passed through the rubber plunger, perpendicular to the shaft of the plunger. The needle and thread are then inserted back down the barrel and out the injection port. The thread is cut off or pulled out of the needle and the two ends are tied together. The knot is then pulled around and back into the injection port and barrel until it rests against the plunger. Next, the plunger can be placed back into the barrel, all the while keeping a loop of thread hanging out of the injection port of the syringe. This loop can then be put around the base of the tail and tightened down by withdrawing the plunger; pushing the plunger down will loosen the tourniquet. Adjustments to the thread length will need to be made based on both the size of the syringe and the desired loop size (Fig 3).26

The tail should be aseptically prepared for sample collection. Alcohol can be used to disinfect the tail, and it should be applied one third to one half of the distance from the tail tip. Holding the tail at this site with the thumb and index finger of one’s nondominant hand will stabilize the section of tail from which blood is to be collected. A 25- to 27-gauge needle alone or attached to a 0.5- to 1-mL syringe is inserted into the skin parallel to the vein and then passed into the vein for 2 to 3 mm (Fig 4). The plunger is withdrawn very gently to reduce the chance of venous collapse. A 25-gauge butterfly needle, with the tubing cut off 2 to 5 mm from the hub of the needle can also be used, with the blood being allowed to drip into a microtainer. If there is a problem collecting the initial sample, another attempt, 0.5 cm cranial to the original site, may be made. Up to 50 μL to 0.2 mL can be safely collected from the mouse tail vein. Hemostasis is performed with light pressure over the site with dry gauze as described above. The lateral tail vein should not be squeezed or milked, because this technique can result in a leukocytosis by mixing the margined white blood cells with the sample or can contaminate the sample with tissue fluids.

Historically, collecting a blood sample from the lateral tail vein has been performed on nonanesthetized animals with a needle or a scalpel blade to cut the vein and then collect the blood into a hematocrit tube or microtainer. This laboratory technique should not be attempted on mice that present to a veterinary hospital. If this procedure is ever used, a topical anesthetic cream should be applied to the tail 30 minutes before the blood sample is collected. The animal should be placed into a restraint tube as described for the saphenous vein and the tail pulled out instead of the rear leg. Lacerating the lateral tail vein will likely produce a sample that is a mixture of tissue fluid and venous or venous and arterial blood.

Ventral Tail Artery. Blood collection from the ventral tail artery is similar to that described for the lateral tail vein, except that the vessel being used is the single ventral tail artery. The tail should be warmed as described above to increase blood flow. Up to 0.5 to 0.75 mL of blood can be collected from the tail artery. One should avoid squeezing or milking the tail during sampling, because it may collapse the artery and slow or impede blood flow. In addition, a blood sample collected from a tail that has been squeezed is more likely to contain a mixture of tissue fluid and arterial and venous blood. Direct pressure can be applied to achieve hemostasis, although pressure will need to be applied for a longer period of time because the sample site is an artery.

General anesthesia should be used for blood collection from the ventral tail artery; however, if there are concerns regarding the risk of anesthesia to the patient, a modification of this technique may be used that does not require a restraint tube or tail.
Thirty minutes before the procedure, the site for sample collection should be cleaned and a topical anesthetic cream (e.g., EMLA cream) applied. The mouse should be placed on a smooth table and light pressure applied to the animal’s back with the lateral edge of the nondominant hand and the last finger. The mouse’s tail should be held with the thumb on the dorsal part of the tail and the index finger on the ventral aspect. The tail is pulled straight up perpendicular to the table. The base of the tail can be stabilized by placing the second and third fingers below the thumb on the dorsal surface of the tail. A scalpel blade is then used to cut across the ventral tail artery at a slight oblique angle to the vessel midway down the length of the tail. Blood can be collected in a microtainer or microhematocrit tube, after which light pressure is applied to the incision to control bleeding. If subsequent samples are needed, they can be collected more proximately on the tail. In one study, 5 blood samples were collected for plasma corticosterone levels by this method every 2 to 3 days for up to 9 days, and the plasma corticosterone levels did not significantly vary over the sampling period. Histological changes at the sampling sites showed minimal inflammation and rapid healing.

**Venipuncture Sites of Last Resort in Private Practice**

**Orbital Sinus.** There are numerous associated health risks when blood is collected from the orbital sinus of a mouse, which increase if the animal is not anesthetized. The health risks include orbital bleeding with increased pressure on the back of the eye and associated pain, infection, blindness, corneal ulceration, punctured or ruptured globe, keratitis, pannus formation, microphthalmia, proptosis of the globe, panophthalmitis, and fractures of the orbital bones. When this venipuncture method is performed by highly skilled technicians in a research setting, it can be done with few complications and minimal stress to the animal; however, it is rare nowadays for research facilities to use the orbital sinus to collect blood from mice.

Anesthesia is recommended when collecting blood from the orbital sinus. A drop of topical ophthalmic anesthetic solution (Proparacaine Hydrochloride Ophthalmic Solution, 0.5% USP; Bausch & Lomb, Inc., Tampa, FL USA; Tetracaine Hydrochloride Ophthalmic Solution USP, 0.5%; Bausch & Lomb, Inc.) should be applied to the surface of the eye and any excess removed after 5 to 10 seconds with dry gauze or a cotton swab. The animal should be placed in lateral recumbency on a table or held in the palm of the nondominant hand so that its head is pointing downward. The index finger should be placed above the eye and the thumb below the eye to pull the skin away from around the globe. Using a microhematocrit tube or a fine-walled (1-2 mm outside diameter) borosilicate glass Pasteur pipette, insert the tip in the corner of the eye socket at the medial or lateral canthus (Fig 5). The tip should be directed toward the middle of the eye socket by directing the tip at a 30° to 45° angle to the side of the head. The tube should be rotated while applying gentle downward pressure until blood is seen in the tube. Once blood is observed in the tube, slightly withdrawing it will increase the blood flow from the sinus. Once the blood stops flowing, the tube is removed and the eyelids are pulled together and pressure is applied to the globe. The skin around the eye should be wiped with dry gauze to remove any blood, being careful not to touch the cornea. No ophthalmic ointment should be applied, because it may cause the animal to rub its eye. The mouse should be monitored for 30 minutes for swelling and/or bleeding from the collection site. Up to 0.2 to 0.3 mL of blood can be safely collected from this site, but it is important to recognize that the sample is a mixture of blood and tissue fluid.

Before use, the microhematocrit tube should be checked for any rough edges that could increase tissue injury around the globe. When using the Pasteur pipette, one should cover the open end of the pipette with a finger before removing it from behind the eyeball to prevent blood from dripping out. If blood is collected from the orbital sinus, at least 21 days are required between bleedings from the same

![Figure 5](image-url). Orbital sinus bleeding in a mouse. A microhematocrit tube is inserted in the medial canthus of the eye. The microhematocrit tube is rotated while applying gentle downward pressure, and the tip is directed toward the middle of the eye socket by directing the tip at a 30° to 45° angle to the side of the head. Once the blood is seen in the tube, it should be withdrawn slightly to facilitate filling.
eye. Blood collection should be alternated between the two eyes and done no more than twice on each eye.

**Tail Clipping, Tail Cut Bleeding.** These aggressive bleeding techniques should be done with the animal under general anesthesia and only if all other sites are unavailable. It is generally recommended that these techniques only be used for terminal patients because of the associated postrecovery pain. The tail tip is prepared aseptically and a topical anesthetic cream (e.g., EMLA cream) is applied 30 minutes before blood collection. The tail should be warmed as previously described to facilitate vasodilatation. A tourniquet is placed around the tail base, and the distal 1 to 2 mm of the tail is amputated perpendicularly to the axis of the tail with a scalpel blade or sharp scissors. Approximately 10 µL of blood can be collected in a capillary tube. The tail tip bleeding collected is a mixture of venous, arterial, and tissue fluids. The collection site should be held off with dry gauze for 30 to 45 seconds. Only the fleshy part of the tail tip should be cut and not any skeletal structures (e.g., caudal vertebrae and/or anterior cartilage). If no blood is observed when the first cut is made, another cut should be made 2 to 3 mm proximal to the initial attempt. For a serial blood sample collection, the scab or blood clot can be removed as needed, with no more than 4 samples being collected over a 24-hour period.

**Toe Clipping, Nail Clipping, Ear Clipping.** Toe clipping, nail clipping, and ear clipping are not acceptable in veterinary hospitals and will probably not produce diagnostic samples.

**Dorsal Pedal Vein, Lateral Marginal Veins of the Tarsus.** The mouse should be placed under general anesthesia for these blood collection techniques. The mouse is warmed to increase its body temperature as described above. The hind foot is held at the ankle by placing the thumb on the top of the foot and the index finger on the plantar aspect. The area is aseptically cleaned and petroleum jelly or silicone grease is applied over the vessel. A 25- to 27-gauge needle is placed in the vein and blood is collected from the hub of the needle in a microhematocrit tube, or the vein is nicked with a 25- to 27-gauge needle and blood is collected in a microhematocrit tube. If the latter technique is used, then a topical anesthetic cream (e.g., EMLA cream) should be applied to the foot 30 minutes before the procedure. Hemostasis can be achieved by applying direct pressure with dry gauze to the venipuncture site. If general anesthesia is not available for the procedure, the animal can be placed in a restraint tube and sampled with the technique described for the saphenous vein. The mouse may show signs of lameness after this blood collection procedure.

**Venipuncture Sites for Terminal Blood Collection**

**Axillary Region.** For blood collection from the axillary region, general anesthesia must be used. Once anesthetized, the mouse should be placed in dorsal recumbency with the front leg abducted laterally. A scalpel blade or sharp scissors should be used to incise the skin over the axillary region. The skin at the caudal edge of the incision should be lifted with a forceps to form a pocket. The axillary vessels should be incised and the blood, up to 0.8 mL, collected with a microhematocrit tube or microtainer. The blood sample collected from the axillary area will contain a mixture of venous and arterial blood and tissue fluids. After collecting blood from the mouse, it should be humanely euthanized.

**Laparotomy or Thoracotomy Sites.** After placing a mouse under general anesthesia, one can collect blood from the posterior vena cava through a laparotomy incision or from the aorta or heart via a thoracotomy. The laparotomy procedure is initiated by incising the skin 1 cm caudal to the rib cage. Once in the body cavity, the widest part of the posterior vena cava can be located between the kidneys after displacing the intestines to the left and the liver in a cranial direction. Blood is collected with a 23- to 25-gauge needle attached to a 1-mL syringe. Up to 0.8 mL of blood can be collected, after which the mouse should be humanely euthanized. Blood can also be collected from the aortic arch and the heart via a thoracotomy.

**Cardiocentesis.** Cardiocentesis for blood collection is a terminal procedure only and must be done with the animal under general anesthesia. The mouse should be placed in dorsal recumbency for the procedure. A 22-gauge, 0.02- to 0.04-inch (0.5-0.9 mm) needle on a 1-mL syringe can be used for the procedure. The needle should be inserted slightly left of midline under the xiphoid cartilage at a 20° to 30° angle from the horizontal axis created by the sternum. While advancing the needle toward the heart, apply slight negative pressure until blood enters the hub to confirm that the heart has been penetrated. Another position from which cardiocentesis can be achieved is by holding the mouse vertically by the fur on the nape of the neck. A 22-gauge, 1-inch needle can be inserted 0.5 cm cranial to the center of the thorax and directed at a 25° to 30° angle cranially. Blood will be seen in the hub of the needle after the needle penetrates approximately 5 to 10 mm into the thoracic cavity. A third method for cardiocente-
sis involves placing the mouse in lateral recumbency so that the needle can be directed perpendicular to the chest wall at the point of maximum intensity of the heart beat. The heart can also be approached via the thoracic inlet with a 25-gauge, 0.02-inch (0.6-mm) needle on a 1-mL syringe. The mouse should be held in the palm of the nondominant hand in dorsal recumbency with the thumb and the index finger holding the scruff of the neck and the last 2 fingers holding the tail. The needle should enter the heart when the hub of the needle passes the lower lip of the mouse. The animal’s head should be pulled down slightly, and the head and body must be held in a straight line. The needle can be directed into the thoracic inlet by lining it up against the mouse’s left cheek. The needle and syringe need to remain parallel to the ventral horizontal plane of the mouse. Gentle negative pressure should be applied to the syringe until blood fills the needle hub. If blood does not appear, pull back on the needle without removing it from the chest and redirect the needle at a slightly different angle. If the blood stops flowing into the syringe, try pulling the needle out slightly or rotating the needle. Up to 0.8 to 1 mL of mixed arterial and venous blood can be collected from the heart. The mouse should be humanely euthanized after the cardiocentesis. Only under extreme circumstances should blood collection via cardiocentesis be performed on a nonterminal patient, then the site of needle entry must be shaved and surgically prepared. As stated before with critical bleeding techniques, the mouse must be maintained under general anesthesia.

**Rat (Rattus norvegicus)**

Rats are susceptible to stress through transport (which can decrease corticosterone levels for up to 3 days), restraint, and introduction into a new habitat. Psychological stress in the rat can cause decreased blood glucose, urea, and fatty acids, and an increase in cholesterol. Stress can also lead to increased corticosterone levels, which have been shown to peak after 15 minutes. A rat can communicate stress with conspecifics through pheromones and by vocalizations; this has not been identified in mice. Therefore, rats that are not being handled but are housed near other rats that are being bled or euthanized may have adversely affected test results.

The previous comments describing how to anesthetize a mouse with minimal stress are also applicable to the rat. Often, the pet rat is far more tractable than the pet mouse because they are handled more frequently by their owners and, in general, have very good dispositions. Therefore, some of the procedures listed below may be possible without anesthesia. For rats that are fractious, isoflurane anesthesia is highly recommended for blood collection. Rats that are ill and may be compromised by handling, should be anesthetized with isoflurane and should be very closely monitored before, during, and after the procedure.

**Venipuncture Sites Recommended for Private Practice**

**Saphenous Vein.** If the rat is tractable, then it may be wrapped in a towel or put into the appropriate-sized syringe case (50 mL) with holes in the tip as described for the mouse. A topical anesthetic cream (e.g., EMLA cream) should be applied to the leg 30 minutes before collecting the sample to reduce the likelihood of the animal pulling on the rear leg when the procedure is performed. Using a 22- to 23-gauge needle on a 1-mL syringe, the needle is directed at a 20° to 30° angle to the vein, or the needle is bent at the hub by 20° to 30° to enter the vein. One should not perform this procedure more than 3 times on an awakened rat, or the stress of handing will affect the diagnostic test results.

**Femoral Vein.** This blood collection procedure in rats is similar to that described for the mouse.

**Jugular Vein.** Blood collection from the jugular vein in the rat is similar to that described in the mouse. The animal should be induced and maintained under general anesthesia and then placed in dorsal recumbency. The head and neck should be hyperextended with a strip of gauze or a piece of string around the upper incisors. When performing this procedure, it is important to closely monitor the patient’s respiration. The neck is prepared as described for the mouse, and a 23-gauge, 1-inch needle on a 1- to 3-mL syringe is inserted through the pectoral muscles just cranial to where the external jugular passes between the clavicle and the pectoral muscle. The use of a sloped restraint board will facilitate the collection of up to 2 mL of blood from an anesthetized rat. In a study by Toft and coworkers, rats anesthetized with isoflurane for jugular, retro-orbital plexus, and tail venipuncture and monitored for heart rate, blood pressure and body temperature were found to recover more quickly from jugular venipuncture than the other techniques, leading the authors to recommend jugular venipuncture over the other methods.

**Dorsal and Lateral Tail Vein.** Rats have a dorsal tail vein and 2 lateral tail veins. To collect blood from the tail veins of a rat, the patient can be manually restrained, placed under general anesthesia, or held in a restraint tube. In a study by Fluttet and coworkers,
ers, rats were preconditioned for 2 minutes a day for 4 to 5 days before blood collection from the dorsal tail vein. The rats were handled and allowed to crawl under a folded towel, which limited their movement and simulated a tunnel created by hands. The tail was gently pulled out from the towel and stroked during this time. On the day of blood collection, the end of the tail was held on the table between 2 fingers, and a 2-mm incision was made in a diagonal direction about 15 mm from the tip of the tail on the dorsal surface of the tail. The fingers above the incision applied more pressure at the time the incision was made. The blood was collected with a microhematocrit tube, and, if needed, the tail was stroked gently from the base to the site of the incision until the total blood volume (up to 300 \( \mu L \)) was collected. The corticosterone levels of these animals remained low (< 2 \( \mu g/dL \)) if the animals were returned to their original enclosures. The venipuncture technique for the lateral tail veins of rats is similar to that described for mice. A 21- to 23-gauge butterfly extension set can be used with the rubber tubing cut off 5 mm from the hub of the needle or the hub of the needle with blood being collected as it drips out into a microtainer tube or aspirated into a 22-gauge needle on a 1-mL syringe.

In a study by Toft and coworkers, rat tail venipuncture was associated with body temperature fluctuations for >30 hours post sampling. The authors suggested that the stress of prolonged handling could interfere with the thermoregulatory mechanisms of the rats. In another study conducted in unanesthetized rats, blood corticosterone and adrenocorticotropic hormone levels remained at baseline levels if the blood collection procedure was completed in <2 to 3 minutes, but if sampling took longer, the levels of the monitored hormones showed a significant elevation.

**Ventral Tail Artery.** General anesthesia should be used when collecting a blood sample from a rat’s ventral tail artery. The rat should be placed in dorsal recumbency for the procedure and the venipuncture site should be aseptically prepared. The preferred venipuncture site is approximately one third of the distance from the tail base. A 19- to 27-gauge needle attached to a plunger-less syringe can be used to collect the sample. The needle should be inserted bevel up at a 30° angle on the ventral midline of the tail. A “pop” should be felt as you enter the artery. Blood can be collected as it drips from the syringe and placed into a microtainer tube. Approximately 0.2 to 3 mL of blood can be safely collected from the rat (based on weight).

**Cranial Vena Cava.** Rats should be placed under general anesthesia for cranial vena cava venipuncture to minimize the risk of thoracic trauma or hemorrhage, and puncture or laceration of the heart, trachea, or jugular vein. For the procedure, the rat should be placed in dorsal recumbency with the forelegs pulled back along the sides of the chest. A 23- to 25-gauge needle on a 1- to 2-mL syringe should be inserted 3 to 8 mm lateral to the manubrium and just cranial to the first rib. The needle is then directed toward the opposite femoral head at a 30° angle down towards the table that the animal is lying on. The needle may need to be advanced 2 to 10 mm to collect the sample. The volume of blood that can be collected from this site varies (0.8-2.5 mL). Once the needle is withdrawn, pressure should be placed on the collection site for up to 30 seconds to ensure proper hemostasis.

**Venipuncture Sites of Last Resort in the Private Practice**

**Dorsal Metatarsal Vein, Interdigital Space.** The leg of the patient should be held at the stifle joint for the leg to be extended and the blood vessels to be held off. The fur over the foot should be clipped and the site prepared aseptically. A 27-gauge or smaller needle can be placed in the vein and the blood collected from the hub of the needle with a microhematocrit tube. Another method has been described in which a 20-gauge needle is used to puncture the interdigital space of the hind foot of an anesthetized rat to collect between 0.5 and 1.0 mL of blood.

**Orbital Plexus.** The orbital plexus is not a suitable site to collect blood from the rat, as this anatomic structure lies deep within the orbit. In one study, bleeding from the orbital plexus in rats was associated with increased serum creatine kinase levels at the time of blood collection. The increased serum creatine kinase was presumed to be associated with the tissue damage that occurred when collecting the samples. A prolonged (up to several weeks) reduction in circulating white blood cells due to a possible stress-induced lymphopenia was also noted along with increased prolactin and corticosterone levels. In another study, serum levels of lactate dehydrogenase, aspartate aminotransferase, malate dehydrogenase, pyruvate kinase, and myokinase were increased in samples collected from the orbital plexus compared with samples collected from the jugular vein.

**Sublingual Vein.** The risks associated with blood collection from the sublingual vein are bruising and swelling of the tongue, which can lead to anorexia. The sublingual vein is not a suitable venipuncture
site for rat patients presented to veterinary hospitals. If used, the rat should be placed under general anesthesia, the tongue extended from the mouth, and a 23- to 26-gauge needle inserted into the vein for sample collection. Blood can be collected directly from the hub of the needle. Zeller and co-workers reported that there was an initial drop in the white blood cell count that lasted for 2 hours post blood collection from the sublingual vein, after which the white blood cell count increased slightly above baseline.

**Tail Clipping, Toe Clipping.** Tail and toe clipping are not acceptable venipuncture methods for companion rat patients.

**Venipuncture Sites for Terminal Blood Collection**

**Laparotomy or Thoracotomy Sites.** These surgical procedures are performed with the same techniques described for the mouse, with blood being collected from the abdominal aorta, caudal or dorsal aorta, abdominal vena cava, or hepatic portal vein. The procedure must be done with general anesthesia. A 19- to 20-gauge needle can be used to collect the sample. Up to 5 to 15 mL of blood can be safely collected from a rat with these techniques. The animal must be humanely euthanized after the procedure.

**Translumbar Vena Cava.** The rat translumbar vena cava is located to the right of the dorsal midline and runs parallel to the aorta. The rat should be anesthetized and placed in sternal recumbency for this procedure. The person collecting the sample should hold the rat with his or her nondominant hand so that the index finger and thumb can be placed on either side of the rat behind the last rib. Holding the animal in this position will stabilize it and allow the phlebotomist to verify the site of the needle’s entrance, which should be at the level of the first lumbar vertebrae. A 25-gauge, 5/8-inch needle should be inserted through the paraspinous muscles 1 cm off the right side of midline and inserted in a coronal plane at an angle of 45° from the vertical. The needle should be inserted until it contacts the transverse process of the first lumbar vertebrae. The needle is then slightly withdrawn and redirected at a more acute angle to miss the bone. The site of the collection is caudal to the diaphragm and cranial to the renal arteries. The risks associated with this venipuncture technique include lacerating the translumbar vena cava or aorta and a 10% to 30% incidence of sample hemolysis. The animal should be humanely euthanized after the blood sample (2 mL) is collected.

**Cardiocentesis.** The approach used for cardiocentesis in rats is similar to the techniques described for mice. With the animal under general anesthesia, a 23-gauge, 1- to 1.5-inch needle on a 1- to 15-mL syringe is used and 5 to 15 mL of blood can be collected. If the patient is not being euthanized, only 1 mL of blood should be collected.

**Guinea Pig (Cavia porcellus)**

Because guinea pigs have very small peripheral veins, short legs and neck, and a compact body, venipuncture in these animals can be challenging. To increase your success with venipuncture in a guinea pig, the patient should be warmed in a 104°F (40°C) incubator to dilate the veins and then placed under general anesthesia. It is important to note that guinea pigs do have a longer prothrombin time than that of rats, rabbits, and hamsters; therefore, their blood will not clot as quickly as that of the other animals mentioned.

**Venipuncture Sites Recommended for Private Practice**

**Ear Veins.** The ear veins of guinea pigs can be nicked with a 25-gauge needle, and the subsequent blood droplets can be collected into a microhematocrit tube.

**Saphenous Vein.** The lateral saphenous vein of the guinea pig runs dorsally and then laterally over the tarsus. The veins are very small; therefore, to visualize the venipuncture site the fur will need to be shaved and alcohol applied over the vein. The blood sample is collected with a 1-mL syringe on a 23- to 27-gauge needle. Pressure should be placed on the thigh to extend the leg and to hold off the vein as described for the rat. In addition to using the needle and syringe, blood may be collected from the hub of the needle with a hematocrit tube.

**Cephalic Vein.** Blood is collected from the guinea pig cephalic vein using the same methods described for the cat.

**Jugular Vein.** Guinea pigs have necks that are very short, which can make this procedure difficult. The animal will need to be anesthetized to be properly restrained for the blood collection in order to eliminate the stress of restraint. The fur will need to be shaved in order to visualize the vein. For proper positioning and optimal exposure of the jugular vein, the cavy’s head should be extended and the forelegs pulled down over the edge of a table while the animal is in sternal recumbency. It is important not to overextend the patient’s head and neck and to monitor the animal’s respirations. If the guinea pig has difficulty breathing, then the procedure...
Blood Collection in Small Mammals

should be aborted. The jugular veins lie within the superficial fascia of the ventral cervical area, with the right jugular being larger than the left. If the jugular veins cannot be visualized or accessed with the animal in ventral recumbency, then the animal can be put in dorsal recumbency for the procedure. An assistant holds the left front leg and the hind limbs, while the individual collecting the blood from the right jugular vein should hold the head of the animal with the thumb between the rami of the mandible and the right foreleg with the fingers. Using a 24-gauge, 5/8-inch needle on a 1-mL syringe, the needle should be directed into the hollow of the right shoulder above the clavicle and toward the left hip. The vein is very superficial, so as the needle is advanced, the plunger should be withdrawn to create negative pressure within the syringe. Once blood starts to fill the syringe, the needle should not be moved to reduce the incidence of cardiac puncture. A blood sample of up to 2.5 mL can be collected from the jugular vein.

Another method used to collect blood from guinea pigs is “sternal notch phlebotomy.” This technique requires general anesthesia with the animal in dorsal recumbency and positioned as symmetrical as possible. A 25-gauge butterfly catheter can be used to collect the sample. The needle should be inserted at about a 20° angle to the body from the left shoulder toward the right hind leg to collect the sample. The blood sample is collected from one of several vessels in this area including possibly the jugular or the cranial vena cava.

**Femoral Vein.** To collect blood from the femoral vein, the guinea pig should be anesthetized and placed in dorsal recumbency. The rear leg to be used for the guinea pig should be anesthetized and placed in dorsal recumbency. To collect blood from the femoral vein, the cranial vena cava. Femoral Vein.

**Venipuncture Sites of Last Resort in Private Practice**

**Interdigital Blood Collection.** This procedure requires that the guinea pig is anesthetized. A topical anesthetic cream (e.g., EMLA cream) should be applied between the digits 30 minutes before the blood is collected. To collect the sample, the digits should be spread apart, excess fur should be removed, and the skin of the interdigital area should be thoroughly prepared using aseptic technique. A 20-gauge needle or a 22- to 23-gauge needle on a 3- to 6-mL syringe should be used to collect the blood. The optimum site for femoral vein blood collection is where the body wall meets the upper thigh, just behind the inguinal nipple and midway along the upper thigh. The needle should be directed perpendicular to the skin or at a 45° angle to the long axis of the femur and inserted into the tissue about 6 mm (0.25 inch) while gently withdrawing on the plunger. Up to 3 mL of blood can be collected from the femoral vein. With this venipuncture technique there is a possibility that the sample could be arterial given the close proximity of the femoral artery to the vein as it runs parallel to the vein and slightly in front of the vein. If bright red blood is collected, be sure to apply pressure to the venipuncture site for a minimum of 2 minutes.

**Cranial Vena Cava.** Guinea pigs should be anesthetized for cranial vena cava venipuncture. The patient should be placed in dorsal recumbency with the forelegs pulled back to the side of the chest or pulled forward. It is important that the animal is positioned squarely on the table in perfect alignment. The landmarks for needle insertion are the manubrium and first rib. A 26-gauge butterfly or a 22- to 23-gauge needle on a 3- to 6-mL syringe should be used to collect the sample. The needle should be directed into the sternal notch under the first rib at a 20° to 35° (depending on the side of the guinea pig) angle toward the right hind leg. The needle should only be inserted 1 to 1.5 cm into the thoracic cavity, while maintaining a slight negative pressure to the syringe. The heart lies very cranial in the thoracic cavity of the guinea pig, and there is a risk of thoracic hemorrhage if the needle punctures cardiac tissue. If blood is not aspirated into the syringe, the phlebotomist should pull the needle out slightly and redirect slightly toward the midline of the anesthetized patient.

**Orbital Sinus.** The approach to orbital sinus venipuncture in guinea pigs is similar to that described for mice, although in guinea pigs the approach is through the lateral canthus. In animals larger than mice, one can use a small-diameter polyethylene tubing in which a bevel cut has been made on the end that will be used to enter the lateral canthus. This causes less eye trauma and has a lower incidence of epistaxis.

**Venipuncture Sites for Terminal Blood Collection**

**Cardiocentesis.** The guinea pig is anesthetized and positioned in dorsal recumbency for cardiocente-
sis. A 20- to 25-gauge, 1-inch needle can be used to
to enter the heart as described in the mouse. This
should only be done on animals that will be hu-
manely euthanized immediately after blood
collection.

**Laparotomy or Thoracotomy Sites.** Blood can be col-
lected from the same vessels using the surgical pro-
cedures described for the mouse. Up to 10 to 15 mL
blood can be collected from the abdominal aorta,
caudal or dorsal aorta, or the vena cava.

**Translumbar Vena Cava.** Blood collection from a
guinea pig’s translumbar vena cava is performed in a
similar manner to that described for the rat.39

**Syrian or Golden Hamster (Mesocricetus
auratus), Siberian or Djungarian Hamster
(Phodopus sungoris), Chinese or Striped
Hamster (Cricetulus griseus)**

**Venipuncture Sites Recommended for Private
Practice**

**Lateral Saphenous Vein.** The approach to the hamster’s
lateral saphenous vein is similar to the mouse, but a 20-
to 25-gauge needle is used to collect the blood.

**Cephalic Vein.** The hamster should be placed under
general anesthesia when collecting blood from the
cephalic vein. A tourniquet should be placed around
the leg proximal to the elbow. The approach to the
cephalic vein is similar to that of the cat, although a
25-gauge needle is inserted into the vein and the
blood is collected from the hub of the needle.

**Cranial Vena Cava.** Blood collection from the cranial
vena cava of hamsters is similar to that described for
the rat.

**Venipuncture Sites of Last Resort in Private
Practice**

**Orbital Sinus.** To collect blood from the orbital sinus
of a hamster, the patient should be anesthetized.
The technique for collecting the sample is similar to
that described for the mouse. Up to 0.1 to 0.5 mL
of blood can be collected by means of this technique,
but as with the mouse, this procedure is not recom-

**Lateral Vein of the Tarsus.** Blood collection from the
lateral vein of a hamster’s tarsus is similar to that
described for the mouse.

**Sublingual Vein.** Blood collection from a hamster’s
sublingual vein is similar to that described for the rat.

**Ear Vein.** For blood collection from a hamster’s ear
vein, the patient should be placed under general
anesthesia or have an anesthetic cream (e.g., EMLA
cream) applied to the site 30 minutes before the
procedure. Ear vein blood collection is accom-
plished by inserting a needle into the vein and col-
lecting the blood from the hub of the needle.

**Venipuncture Sites for Terminal Blood Collec-
tion**

**Laparotomy or Thoracotomy Sites.** Up to 5 mL (based
on animal size) of blood can be collected through a
laparotomy or thoracotomy incision with a 23- to
25-gauge needle and approaching the procedure as
described for the mouse.

**Cardiocentesis.** Cardiocentesis in the hamster is simi-
lar to the approach described for the mouse and rat.
Up to 5 mL of blood can be collected via cardiac
puncture from the hamster with a 23-gauge needle
with a 1- to 5-mL syringe.

**Translumbar Vena Cava.** Blood collection from a
hamster using the translumbar vena cava is per-
formed as described for the rat.39

**Mongolian Gerbil (Meriones unguiculatus)**
The Mongolian gerbil has a rapid turnover rate for
their erythrocytes, with a 10-day average lifespan for
their red blood cells. Therefore, the response to
blood loss from hemorrhage is much faster in this
species compared with that of other small exotic
mammals. In addition, because of the rapid rate of
erythrocyte replenishment, blood samples can be
collected more frequently without adverse physio-
logic effects in most cases.

**Venipuncture Sites Recommended for Private
Practice**

**Saphenous Vein and Metatarsal Vein.** The gerbil
should be placed under general anesthesia or an
anesthetic cream (e.g., EMLA cream) applied 30
minutes before collecting blood from the saphenous
and/or metatarsal vein. To prepare this site for
blood collection, a rubber band tourniquet should
be placed proximal to the stifle and the leg ex-
tended. Clipping the fur over the vein is also recom-

**Lateral Tail Vein.** Blood collection from the lateral
tail vein of a gerbil is similar to that described for a
mouse, although gerbils are prone to degloving in-
juries of their tail and should be handled carefully.

**Venipuncture Sites of Last Resort in Private
Practice**

**Orbital Sinus.** General anesthesia is recommended
for orbital sinus venipuncture in gerbils. The orbital
sinus of a gerbil should be approached through the lateral canthus. To facilitate sample collection, the head should be stabilized with the thumb and forefinger. Placing the thumb behind the mandible will occlude the external jugular and increase blood flow to the orbital sinus. The microhematocrit tube is directed posteriorly and medially under the eye. Up to 0.1 to 0.3 mL of blood can be safely collected from the orbital sinus of a gerbil.

**Venipuncture Sites for Terminal Blood Collection**

**Cardiocentesis and Laparotomy or Thoracotomy Sites.** Cardiocentesis and laparotomy or thoracotomy incisions for terminal blood collection in gerbils can be done with the same methods described for the mouse.

**African Hedgehog** (*Atelerix albiventris*), **European Hedgehog** (*Erinaceus europaeus*)

Hedgehogs present a real problem to the phlebotomist. In addition to the obvious difficulty of working with an animal with numerous dorsal spines, they also have small peripheral vessels which have a tendency to collapse. General anesthesia can be used to overcome these issues. Hedgehogs may hypersalivate when exposed to isoflurane gas, so atropine may be required. These animals also have a tendency for becoming obese, which can make blood collection problematic.

**Venipuncture Sites Recommended for Private Practice**

**Lateral Saphenous Vein and Cephalic Vein.** Both of these sites can be difficult to sample on a hedgehog patient, although access may be facilitated by increasing the animal’s body temperature. Up to 0.5 mL of blood can be collected from these sites. The approach to the lateral saphenous and cephalic vein of a hedgehog is similar to that described for the hamster. These vessels do have a tendency to collapse when negative pressure is applied to a syringe, which can lead to small volume samples and poor-quality blood samples. To avoid venous collapse, one can collect the blood in the hub of a needle with a microhematocrit tube or a pre-heparinized needle and syringe.

**Jugular Vein.** The jugular vein may be difficult to visualize unless a hedgehog is thin. The approach to a hedgehog’s jugular vein is similar to the procedure described for the cranial vena cava previously. The jugular vein lies halfway between the ramus of the mandible and the point of the shoulder. A 22- to 25-gauge needle that is bent at the hub at a slight 30° angle and fastened to a 1- to 3-mL syringe can be used to collect a jugular blood sample from a hedgehog.5

**Femoral Vein.** The approach to the hedgehog femoral vein is similar to that described for the guinea pig. Less than 1 mL of blood can be collected from the femoral vein of a hedgehog, and special care should be taken not to lacerate the femoral artery.

**Cranial Vena Cava.** The approach to the cranial vena cava of a hedgehog is similar to that of the rat and is the only site from which larger volumes of blood can be collected. A 25-gauge needle on a 1- to 3-mL syringe should be used to collect 1 to 3 mL of blood from the cranial vena cava depending on the size and species of the hedgehog.

**Venipuncture Sites for Terminal Blood Collection**

**Cardiocentesis and Laparotomy or Thoracotomy Sites.** Blood collection with these techniques are performed in hedgehogs similar to those described for the mouse and rat.

**Rabbit** (*Oryctolagus cuniculus*)

A rabbit’s blood vessels are fragile and prone to develop hematomas, so care should be taken when collecting a sample. Rabbit blood clots quickly at room temperature; therefore, it is advisable to pre-heparinize the needle and syringe.45,46 Preloading a syringe/needle with heparin is accomplished by drawing up the anticoagulant into the needle and syringe and then injecting it back into the bottle. Blood samples from rabbits should be placed in ethylenediamine tetraacetic acid for complete blood counts and lithium heparin for chemistry panels. Factors that can adversely affect the results of a rabbit’s hemogram include stress, age, sex, breed, and circadian rhythms.46 When rabbits are handled by strangers, their serum chemistry parameters can be adversely affected. This problem may be minimized if the animal is handled by its owner.46 All of these factors need to be considered when collecting blood and interpreting the results from rabbit patients. Rabbits release high levels of catecholamines when severely sick and stressed, which can lead to cardiac arrest.46 Restraint of a rabbit patient should be minimal and closely monitored, or the animal should be placed under general anesthesia, if possible.

**Venipuncture Sites Recommended for Private Practice**

**Lateral Saphenous Vein.** The approach to a rabbit’s lateral saphenous vein is similar to that previously described for other small exotic mammals, although
restraint is slightly different because of the size of the animal. To restrain a rabbit for lateral saphenous venipuncture, the animal should be placed in sternal recumbency and its head situated between the restrainer’s elbow and body. To gain access to the vein, the rear leg should be pulled over the edge of the table by grabbing at the crux of the stifle, which will both extend the leg and hold off the vein. The rabbit can also be held in lateral recumbency, but two handlers will be needed to restrain the animal in this position: one person to restrain the rabbit’s shoulders and the other the pelvis. The lateral saphenous vein is superficial and is prone to collapse and the development of hematomas, which should be communicated to the client before blood collection.

Cephalic Vein. The rabbit’s cephalic vein is small and has a tendency to roll. A 25-gauge needle on a 1-mL syringe can be used to collect blood from this site following the approach used in cats.

Jugular Vein. The author recommends using general anesthesia for jugular venipuncture in rabbits. However, if the animal is calm, it can be wrapped in a towel or a cat bag for restraint. The biggest concern associated with jugular venipuncture is compromising the animal’s breathing when its head is extended. To limit the likelihood of problems, the animal’s breathing should be monitored closely during the procedure. When a rabbit is in sternal recumbency, the approach to jugular venipuncture is similar to that of a cat. Jugular venipuncture can be difficult in obese animals or in does with a large dewlap. In cases where the dewlap is in the way, it can be displaced ventrally with the phlebotomist’s thumb to help gain access to the jugular vein. A rabbit can also be placed in dorsal recumbency with its head extended for sample collection. The jugular vein can be approached from a cranio-caudal or caudo-cranial direction in relation to the head.

Femoral Vein. Blood collection from the rabbit femoral vein is similar to that described for the guinea pig.

Cranial Vena Cava. The procedure for collecting blood from the cranial vena cava is similar to that described for the rat.

Venipuncture of Last Resort in Private Practice

Marginal Ear Vein. Given the risk of thrombosis and subsequent sloughing of the pinna, especially in small-eared rabbits, the marginal ear vein is not considered a suitable site for venipuncture in rabbit patients. If this site is used for venipuncture, the author recommends anesthetizing the rabbit and applying a topical anesthetic cream (e.g., EMLA cream) 30 minutes before sample collection to prevent the animal from moving and causing subsequent laceration of the vein. A 25- to 26-gauge needle can be used to collect blood from the vein, which tracks along the margin of the ear. Blood collection from this site should be done at the tip of the ear. To better visualize the vein, the patient’s fur should be shaved or plucked, taking care not to tear the delicate skin. To access the ear veins, the ears should be warmed before sample collection. The ears may be warmed with a low-wattage light bulb, one’s hand, or a warm towel. Warm water put into a plastic glove, with the open end tied closed, will not only increase the temperature of the ear but can be placed inside the pinna to support the structure for blood collection. Sedatives such as acepromazine maleate (Acepromazine Injectable; Vedco, Inc., St. Joseph, MO USA) will also aid in dilation of the vessels. The vessels can be engorged by holding the ear at its base. The needle should be inserted through the skin, parallel to the vein, and, once under the skin, directed into the vein. Using this technique will minimize the risk of hematoma and also aid in entering the veins, which have a tendency to roll. Up to 0.5 to 10 mL of blood can be safely collected depending on the size of the rabbit.

Central Ear Artery. A 21- to 22-gauge butterfly catheter can be used to collect blood from a rabbit’s central ear artery (Fig 6). Tension should be applied to the ear to assist with inserting the needle. The needle should be directed toward the base of the ear, with the insertion point closer to the tip of the ear. The author recommends bending the needle at the hub to gain access to the artery. As with the marginal ear vein, the needle should be inserted parallel to the artery and then directed into the vessel once it is in the subcutaneous space. The artery does not require warming for collection; however, digital pressure over the collection site for several minutes will be needed for hemostasis. Depending on the size of the rabbit, up to 30 to 40 mL can be collected safely from this site.

Figure 6. Rabbit ear with central artery and lateral veins identified.
Venipuncture Sites for Terminal Blood Collection

Cardiocentesis. Blood collection from this location will require general anesthesia and should only be used if the animal will be euthanized after the procedure. An 18- to 21-gauge, 1.5-inch needle can be used for rabbit cardiocentesis, and the approach is similar to that described previously. Depending on the size of the rabbit, 60 to 200 mL of blood can be collected from the heart.

Ferret (Mustela putorius furo)

Blood collection in the ferret is similar to that described and used for the domestic cat. Because ferrets are often handled by their owners, the use of anesthesia may not be necessary. If isoflurane is used, the clinician should be aware of its possible effect on the results. In addition, a ferret hemogram should not be used, because it will rapidly elevate blood glucose levels. Isoflurane anesthesia can be used to help facilitate sample collection from ferrets.

Venipuncture Sites Recommended for Private Practice

Lateral Saphenous Vein. To collect blood from a ferret’s lateral saphenous vein, the animal should be placed in ventral or lateral recumbency and the rear leg grasped just distal to the stifle to engorge the vessel. A 26- to 27-gauge needle on a 1-mL syringe can be used to decrease the risk of vessel collapse during the blood draw. A topical anesthetic cream (e.g., EMLA cream) should be applied to the vessel 30 minutes before sample collection to minimize patient discomfort.

Cephalic Vein. Restraint and blood collection from a ferret’s cephalic vein is similar to that used for dogs and cats. The ferret can be wrapped in a towel for restraint or grasped by the scruff of the neck and held in a vertical position. By restraining the patient’s “scruff,” the animal becomes passive and will allow the foreleg to be held and a tourniquet placed above the elbow. A topical anesthetic cream (e.g., EMLA cream) should be applied 30 minutes before this procedure. For cephalic venipuncture, a 25- to 26-gauge needle on a 1-mL syringe is recommended. For the cephalic collection, do not roll the vein as one would in the dog.

Jugular Vein. Blood collection from a ferret’s jugular vein is similar to that of the cat, although the vein lies in a more lateral position. A 20- to 25-gauge needle should be used to collect the sample, and bending the needle at a 30° angle will help facilitate sample collection. The animal can be wrapped in a towel or restrained with the head and neck extended and the forelegs pulled down over the edge of the table. Shaving the fur over the vein and applying pressure at the thoracic inlet will help the phlebotomist visualize the vein. If the patient struggles, it can be calmed by feeding it a liquid treat (Lindane; Church & Dwight, Princeton, NJ USA). A sugar-based treat should not be used, because it will rapidly elevate blood glucose levels. Isoflurane anesthesia can be used to help facilitate sample collection from ferrets.

Cranial Vena Cava.

Ferrets have a very long thoracic cavity, and their heart lies in the caudal aspect of their thorax. Because of the caudal location of their heart, the risk of cardiac puncture with cranial vena cava venipuncture in this species is minimal. To access the cranial vena cava of a ferret, the patient should be grasped by the scruff of the neck and placed in dorsal recumbency. The head should be positioned off the edge of the table, keeping the head and neck in alignment with the body, while the front legs are pulled back along the chest. To prevent rotation of the forelegs, two fingers should be placed between the legs. Another person is required to support the body just cranial to the pelvis. It is not necessary to hold the back legs, and this may be stressful to the animal. A 25- to 27-gauge, 0.5-inch needle on a 3-mL syringe can be used for sample collection. The needle should be inserted into the thoracic notch between the manubrium and the sternum, just cranial to the first rib. Most people recommend directing the needle at a 45° angle toward the opposite hip. However, Wolf recommends that if the animal is being bled from the left side, then the needle should be directed toward the right elbow or right hip. If bled from the right cranial vena cava, the needle should be inserted parallel to the sternum and more superficial to the thoracic cavity. Minimal negative pressure is required within the syringe as the needle is advanced. The ferret can also be wrapped in a towel to restrain the head and forepaws with the procedure performed as described above. If the animal is difficult to handle, general anesthesia should be used for sample collection.

Venipuncture Sites of Last Resort in Private Practice

Orbital Sinus. Although this site is recommended in the literature, it is not suitable for the ferret and there are far better superficial sites that can be used. Periorbital fibrous tissue is very tough in the ferret; therefore, the technique that is needed for blood collection from this site requires that the capillary tube be pushed hard to enter the periorbital sinus in ferrets.

Caudal Artery of the Tail. Blood collection from the caudal tail artery in ferrets appears to be painful for the animal and should be used as a last resort or
when the patient is under general anesthesia. To increase the likelihood of collecting a sample, the animal may need to have its body temperature elevated by placing it in a warm room or applying a warm compress to the ventral tail. If the animal is to be bled from the caudal artery of the tail without general anesthesia, a topical anesthetic cream (e.g., EMLA cream) should be applied to the tail 30 minutes before performing the procedure. The ferret should be placed in dorsal recumbency, and a 20- to 21-gauge needle on a 1- to 3-mL syringe should be inserted into the groove on the midline of the ventral surface of the tail 1 to 2 inches (2-5 cm) from the base of the tail and at a 45° angle toward the body of the animal. The artery is very superficial (2-3 mm deep). Pressure should be applied to the venipuncture site for 1 to 3 minutes to ensure proper hemostasis. Blood test results are reported to vary considerably when blood is collected from the caudal artery of the tail.47

Venipuncture Sites for Terminal Blood Collection

Cardiocentesis. The animal needs to be anesthetized for this procedure. By using a 20-gauge, 1.5-inch needle, the approach for cardiocentesis in the ferret is similar to that described for other small exotic and domestic mammals. The veterinary clinician should recognize that the landmarks for locating the heart are at the sixth to eighth ribs. Up to 20 mL of blood can be collected from ferrets with this technique.

Laparotomy or Thoracotomy. Collecting blood from terminal ferret cases with these surgical techniques is similar to that described for the rat.

Chinchilla (Chinchilla lanigera)

The femoral, jugular, cephalic, lateral saphenous, lateral abdominal, and ventral tail veins can be used to collect blood from chinchillas, with the cephalic and lateral saphenous being the author’s preferred sites. A 23- to 27-gauge needle on a 0.5- to 1-mL syringe is commonly used to collect blood from chinchillas for diagnostic testing. It is important to note that hematology and serum chemistry results in chinchillas can vary depending on seasonal changes (e.g., temperature, changes in photoperiod, reproductive cycle).16

Venipuncture Sites Recommended for Private Practice

Lateral Saphenous Vein, Cephalic Vein, Femoral Vein. The approaches to collecting blood from a chinchilla’s lateral saphenous vein, cephalic vein, and femoral vein are similar to the techniques used for cats and described previously. In most cases, general anesthesia is required to collect samples from these sites in chinchillas.

Jugular Vein. On very tame chinchillas, blood can be collected from the jugular vein with the animal conscious, although it is generally preferred to perform this procedure when the animal is under anesthesia. To collect the sample, the patient should be placed in sternal recumbency with its head and neck extended upward. The animal’s breathing must be monitored during the procedure. The jugular vein is superficial in chinchillas, but is difficult to visualize or feel. A 23- to 25-gauge needle that is bent to a 30° to 45° angle and attached to a 1- to 3-mL syringe should be used to collect the sample.

Cranial Vena Cava. The chinchilla patient will need to be anesthetized for blood collection from the cranial vena cava. The approach to collect blood from the cranial vena cava in chinchillas is similar to that described for the guinea pig.

Venipuncture Sites of Last Resort in Private Practice

Orbital Sinus. Blood collection from the orbital sinus is performed in a similar manner to that described for the mouse.

Transverse Sinus. The transverse sinus is used in laboratory investigations but is not suitable for companion animals. In the chinchilla, the transverse sinus encircles the auditory bullae. To approach this sinus, the fur should be removed from the dorsal aspect of the head near the ear and the area aseptically prepared. The transverse sinus in chinchillas is very superficial, and one can use a 25-gauge, 3/8-inch butterfly catheter with a 1-mL syringe to collect the blood. The needle should be inserted at a slight angle medial to the edge of the auditory bulla approximately 1 to 2 mm under the skin. If blood is not noted in the hub of the needle once negative pressure is applied, then the needle needs to be angled at a slightly steeper angle.2

Venipuncture Sites for Terminal Blood Collection

Laparotomy or Thoracotomy Sites, Cardiocentesis. These sites can be approached in a chinchilla with the same techniques described for the rat.

Sugar Glider (Petaurus breviceps)

Sugar gliders are interesting marsupials that have become popular in the exotic pet trade over the last 2 decades. These animals generally have small peripheral vessels, which can make sample collection challenging. Because of these limitations, general
anesthesia with isoflurane is recommended for collecting blood samples from sugar gliders.

**Venipuncture Sites Recommended for Private Practice**

*Lateral Saphenous Vein, Cephalic Vein, Femoral Vein, Ventral Tail Vein.* The lateral saphenous, cephalic, and femoral veins can be used to collect small volumes of blood (0.1-0.25 mL) from a sugar glider. These veins are superficial and often can be visualized; however, they readily collapse even when using a small-gauge needle (e.g., 27-gauge). When collecting a sample from the ventral tail vein, it is recommended to warm the animal before sample collection to vasodilate the vessel.

*Medial Tibial Artery.* A 0.5-mL blood sample can be collected from the medial tibial artery when using a 25- to 27-gauge needle on a 1-mL syringe. The vessel can be visualized but has a tendency to roll. Because this is an arterial sample, pressure will need to be applied to the vessel longer to achieve appropriate hemostasis.

*Jugular Vein.* The jugular veins of sugar gliders are difficult to visualize. A 25- to 27-gauge needle on a 1-mL syringe can be used to collect a 0.5- to 1-mL blood sample from the jugular vein of a sugar glider. The needle should be inserted midway between the point of the shoulder and the ramus of the mandible.

*Cranial Vena Cava.* The approach to cranial vena cava venipuncture in a sugar glider is similar to the technique described for the guinea pig. A 25- to 27-gauge needle on a 1-mL syringe can be used to collect a 0.5- to 1-mL sample.

**Venipuncture Sites of Last Resort in Private Practice**

*Orbital Sinus.* The approach to the orbital sinus of a sugar glider is similar to that described for the mouse.

**Venipuncture Sites for Terminal Blood Collection**

*Laparotomy or Thoracotomy Sites, Cardiocentesis.* The approach to these procedures in a sugar glider is similar to that described for the rat.

**Venipuncture Techniques for the Degu (Octodon degus)** The saphenous, cephalic, and cranial vena cava veins are the preferred sites for blood collection in a degu. Up to 0.5 to 1 mL can be collected from these sites. When collecting blood from these sites in degus, the approach is similar as that described for guinea pigs. When collecting blood samples from degus, it is required that the patient be under general anesthesia.

**Summary**

Collecting blood samples from exotic small mammals can be challenging. To become proficient with exotic small mammal venipuncture, it is important to develop an understanding of the anatomic locations of the vessels and their associated landmarks, and practice, practice, practice. The veterinary clinician should always be aware of the potential risks associated with blood collection from the smallest of these pet species, especially those that are presenting in advanced diseased states.

The clinician should also be aware of the many factors that can affect the blood results in normal healthy animals. As mentioned above, anesthesia, sex, age, reproductive cycle, circadian rhythm, restraint, stress and even the site of the blood sampling can affect the laboratory results. Assessing the validity of the published normal values can be difficult because often when the data is presented in books or review articles, the parameters listed above are not mentioned. Ideally a set of in-house normal blood work should be developed where the variables can be better controlled. The author recommends that clinicians use anesthesia to minimize the stress of handling of the small prey species that are highly adaptive to have a rapid increase in plasma corticosterone levels when exposed to a stressor such as transport to your clinic. An ambulatory type practice may be an ideal way to work with these pocket pets thereby, minimizing the transport and handling stress. In addition, anesthesia may be indicated for the animal’s level of pain, but this is difficult to assess due to the subtleties of each of the different species’ signs of pain and distress (Table 4). Anything that can be done to minimize these effects should be done. Therefore, the clinician will have to carefully consider the benefits of getting the blood sample versus the risk of the collection procedure on the animal.

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