Rodent Diagnostic Testing

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Abstract

Clinical medicine is an important part of scientific medicine that is all too often neglected when treating rodents and small mammal pets. As with more traditional pets, a progressive diagnostic regimen should include a thorough history, clinical signs, physical examination, and laboratory findings.

Key words: **rodent; diagnostic testing; hematology; urinalysis; infectious disease**

On average, the total blood volume in rodent species is 6% to 8% of body weight. Thinner animals will have relatively larger blood volumes per body mass because of greater surface area. Younger animals, especially newborns, have proportionately larger blood volumes than older animals.[1]

The blood volume of a healthy non-rodent mammal ranges from 10% to 15% of body weight, and up to 10% of the total blood volume may be safely removed at any one time or 1% of body weight.[2] The following guidelines should be considered when larger or more frequent sampling is required[1]:

1) 10% to 15% of total blood volume or 1% of body weight is the maximum amount of blood that should be collected at one time;

2) Blood volume is restored in 24 hours, but erythrocytes and reticulocytes may not return to normal levels for up to 2 weeks. Therefore, the maximum amount of blood should be withdrawn only once every 2 weeks. Monitoring the packed cell volume (PCV) or hemoglobin can help evaluate whether the patient has recovered from blood withdrawal;

3) Removal of up to 10% of total blood volume daily over time is permissible; however, the effects of stress, site chosen, and anesthetic used must be carefully considered;

4) Removal of blood volume equal to 20% of the total blood volume is permissible if replacement fluids are given at the time blood is collected. The blood volume removed should be replaced by an intravenous or intraosseous route with twice the volume of body temperature crystalloid fluids at a slow, steady rate. If fluids cannot be administered intravenously, intraperitoneal or subcutaneous routes are other alternatives;

5) 15% to 25% blood loss results in elevated plasma epinephrine, norepinephrine, and corticosterone concentrations to compensate for a decreased level of plasma glucose concentration; and

6) 20% to 25% blood loss decreases the arterial blood pressure, cardiac output, and oxygen delivery to vital organs leading to hypovolemia and cardiac failure (shock). Muscular weakness, depressed mentation, and cold extremities may also be observed. Immediately after blood collection, always observe the patient for signs of distress or anemia (e.g., rapid breathing, pale color of mucous membranes, depressed mentation, or muscle weakness). Observe mice daily for other problems, such as local trauma, infection, or irritation at the blood collection site.

With modern technology a complete blood cell count (CBC), hematocrit(Hct)/packed cell volume

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1557-5063/08/1701-$30.00

doi:10.1053/j.jepm.2007.12.004

(PCV), and several plasma chemistries may be run on as little as 150 μL of whole blood. Therefore, this level of diagnostic medicine is available to patients as small as 15 g without exceeding the 1% of body weight rule. Small domestic (fancy) pet mice (*Mus musculus*) and dwarf hamsters (*Phodopus sungorus* and *Cricetulus griseus*), the smallest of rodent patients, weigh approximately 25 to 30 g, falling easily into this weight category.

Blood Collection Techniques

Collecting blood from a small rodent is perhaps the greatest barrier to obtain laboratory data for these pets. Both the source and method of blood collection can affect blood parameters. Increases in blood hormone and glucose levels are directly related to stressful methods of blood collection. Mice may be stressed by restraining procedures or by sensing impending danger. Blood should not be collected from the orbital sinus more frequently than once every 2 weeks. The tail, saphenous, toenail, and jugular veins can be used for serial blood collection as often as needed.

Anesthesia for Blood Collection

To minimize discomfort to the animal, anesthesia is recommended when collecting blood from rodent patients. Warming the mouse immediately before blood collection will increase blood flow considerably. Place a lamp over the cage for 5 minutes, or place the cage on a heating pad, on the lowest setting. Take care not to overheat the patient.

Jugular

The jugular vein is the preferred site for sampling these patients in the author's practice. Patients are anesthetized with isoflurane and gently restrained in dorsal or lateral recumbency. With smaller patients, the head may be restrained with a loop of string attached to a gauze square-looped around the upper incisors. The head is pulled dorsally and cranially. The area of the vessel is prepared with aseptic technique, and the vein is occluded at the thoracic inlet. The vein may not be visualized in some species but runs from the manubrium to just below the angle of the jaw in all species. The short neck of a rodent and the depth of the jugular vein, often deep to salivary glands, may make entering the vein from cranial to caudal easier for some practitioners. Withdraw blood slowly to avoid collapse of these small vessels. If the first attempt to draw blood is unsuccessful, withdraw the needle slightly; it may have been placed too deeply. If blood stops flowing, do not continue to draw back on the syringe. The vein may have collapsed, or the needle may have attached to the vessel wall. Rotate the needle slightly, or apply slight pressure on the needle (either above, below, or to the side of puncture site).

Toenail Sampling

Depending on toenail anatomy, small volumes of blood suitable for a CBC may be obtained from cut toe nails in some species (guinea pigs [*Cavia porcellus*], rats [*Rattus norvegicus*], mice, and prairie dogs [*Cynomus ludovicianus*] but not hamsters or chinchillas [*Chinchilla lanigera*]). Even this method of collection is best performed while the patient is under isoflurane anesthesia. The foot and nail are thoroughly cleaned and warmed before cutting the nail above the tip of the "quick," amputating the distal tip of the phalanx. Blood may be collected in a heparinized microhematocrit tube or small heparin or ethylenediamine tetraacetic acid blood tube. The nail may be cauterized with styptic powder, Monsel's solution (ferric subsulfate solution), or silver nitrate.

Blood Collection from the Tail

Blood may be collected from the central tail artery or the lateral tail veins from species with a significant tail mass, including mice, rats, chinchillas, gerbils *(Meriones unguiculatus)*, prairie dogs, and Degus (*Octodon degus*). The patient is anesthetized and restrained in dorsal recumbency with the tail exposed. Dilate the vessels by immersion of the tail in water not exceeding 104°F (40°C). The lateral veins lie immediately beneath the skin on each side of the tail. The central artery is midline on the ventral surface of the tail. The author prefers to use the artery for removal of blood and the veins for injection. With the tail under gentle traction and the needle at a 45° angle to the tail, insert needle into the lumen of the vessel with the bevel facing cranial. Blood is withdrawn slowly. When taking blood from the artery, a 25-gauge needle on a 1-mL syringe barrel (no plunger) may be used and the blood is allowed to flow under its own pressure. Flicking or gentle massaging of the tail toward the needle may help facilitate bleeding. Remove the needle and apply gentle pressure to the site of entry to ensure good hemostasis. PCV and hemoglobin measurements have been reported to be higher in blood collected from the tail vein compared with blood obtained from other sites.[3]

Blood may be collected by "tipping" the tail of mice, although it is not a preferred method and should be reserved as an alternative when other