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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. Copyright 2008 Elsevier Inc. All rights reserved.

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 required1:

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 Therefore, the maximum amount of blood should be withdrawn only once every 2 weeks.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...
(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.  

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
approaches fail. The mouse is anesthetized and warmed. Do not attempt to increase blood flow by rubbing the tail from the base to the tip, because this will result in leukocytosis (increased white blood cell [WBC] count). Using a scalpel or straight-edge razor, quickly remove 2 to 3 mm of the tail. Collect blood in a capillary tube as drops appear. Apply pressure or use tissue glue to stop the bleeding. When several samples are needed within a short time period, the original wound can be reopened by removing the clot. When additional samples are needed at a later date, blood samples can be obtained by removing just 2 to 3 mm of additional tail. Cutting the tail too short may result in trauma to the cartilage and ultimately the coccygeal vertebrae.

**Saphenous Vein**

Anesthetize the mouse with isoflurane or other suitable gas anesthetic. Grasp the fold of skin between the tail and thigh. The saphenous vein is found on the caudal surface of the thigh. Remove hair from the area with clippers. Apply petroleum jelly or eye lubricant to prevent migration of blood into the surrounding hair, and place a tourniquet around the leg above the knee. Puncture the vein with a 25-gauge needle. Collect drops of blood as they appear into microhematocrit tubes. Remove tourniquet and apply pressure or apply a drop of tissue glue to stop the bleeding.

**Blood Collection from the Orbital Sinus**

Orbital sinus bleeding, although not aesthetically pleasing, is effective and may be more humane than tail cutting in mice and safer than cardiac samples in mice and hamsters, where obtaining a sample may be very difficult (especially when mice or hamsters are obese). This technique is regularly used in laboratory medicine, where it has been extensively studied; both sample quality and humane issues have been addressed.

Lay the anesthetized patient on its side on a table or a flat surface. With your first finger and thumb (finger above and thumb below the eye), pull the eyelids away from the eyeball, above and below the eye, so that the eyeball is protruding out of the socket as much as possible. Take care not to occlude the trachea with your thumb. Insert the tip of a fine-walled microhematocrit blood tube into the caudal area of the conjunctiva, directing the tip at a 45° angle toward the middle of the socket. Rotate the pipette between your fingers; do not move it from side to side or front to back. Apply gentle downward pressure, and then release until the vein is broken and blood is visualized entering the tube. When a small amount of blood begins filling the pipette, withdraw slightly and allow the pipette to fill. Place the tip of your finger over the end of the tube before removing it from the orbital sinus to prevent spilling. Bleeding usually stops immediately and completely when the tube is removed; however, it may be necessary to apply gentle pressure on the eyeball for a brief moment by closing the skin above and below the eye with your first finger and thumb. Sample collection should not be repeated on the same eye for at least 2 weeks. Blindness can occur if the optic nerve is damaged as a result of the tube coming into contact with that anatomic structure. Corneal ulceration, puncture wounds, loss of vitreous humor, infection, or keratitis may occur as a result of poor technique or uncontrolled movement of the animal.

**Submandibular Blood Collection in Mice**

Blood collection from the submandibular vein is an alternative to retro-orbital blood collection in mice. It has been described in the conscious animal; however, isoflurane anesthesia is recommended. Anesthesia is induced, the hair is shaved from an area below the mandible to even with the ear, and a light layer of petroleum jelly or triple antibiotic eye ointment is applied to the area. The mouse is restrained with the nondominant hand by grasping the loose skin between the ears, extending to the shoulder area. The skin should be taut over the mandible. The submandibular vein is punctured between the mandible and the ear canal with a 25- to 22-gauge needle in a swift motion with only the bevel of the needle. Blood flows immediately if the vein is cut. Blood is collected into a pipette or hematocrit tubes. After the sample has been collected, pressure is applied with a gauze sponge until bleeding has stopped. The mouse should be observed for several minutes to assure that hemostasis has occurred.

**The Complete Blood Cell Count**

The CBC is the most important diagnostic tool available to the veterinarian next to a good history and physical examination. Further, it is of great importance that the CBC be performed by a veterinarian or technician who has a thorough knowledge and experience with the species they are examining.

**Erythrocytes**

Erythrocyte size and morphology vary among true rodents (rats, mice, gerbils, and hamsters) and hysticomorph rodents (guinea pigs and chinchillas). Guinea pigs have large erythrocytes compared with
that of other rodents. Howell-Jolly bodies are common in small numbers of erythrocytes in rats and mice, and Rouleaux formation of erythrocytes is rarely seen, even with inflammation.

The erythrocytes of rodents have a relatively short half-life (45-68 days) compared with that of larger domestic mammals—cat, dog, horse, and cow: 70, 120, 145, and 170 days, respectively. Polychromasia is commonly observed and is related to the short erythrocyte half-life. Adult animals will also have a greater degree of reticulocytosis (2%-7%). Polychromasia and hypochromasia may be used to evaluate the cause of anemia in rodents. When polychromasia is increased in an anemic animal, anemia is often related to blood loss. When polychromasia is not present or if cells are hypochromatic, anemia may be related to erythroid hypoplasia or aplastic anemia. Hypochromasia may indicate a state of iron deficiency related to blood-sucking parasites or inadequate dietary iron.

Erythrocyte and hemoglobin levels of the guinea pig and the rabbit are similar, whereas values are higher for rats, mice, and hamsters. There is no significant difference between the male and female guinea pig. Mean Hct was 2% lower in female rats than males, and plasma volume (mean ± 1 standard deviation) was 4.86 ± 0.54 mL/100 g in females compared with 4.12 ± 0.32 mL/100 g in males.

Packed Cell Volume
PCV or Hct of the guinea pig ranges from 37.0% to 48.0%. At birth, their PCV is higher (48% ± 1.0) than the adult, dropping to the adult range over the first 3 weeks. Hemoglobin of the rat, rabbit, and guinea pig has a relatively high affinity for oxygen, requiring a cofactor to sufficiently lower oxygen affinity for physiological oxygen unloading. Hemoglobin of the guinea pig, rat, mouse, gerbil, and hamster is exceptionally resistant to oxidation by nitrites as opposed to the susceptibility of rabbit, dog, and human hemoglobin to methemoglobin formation.

Leukocytes
As with other mammals, rodents have both granular (neutrophils [heterophils], eosinophils and basophils) and agranular (mononuclear) leukocytes (monocytes and lymphocytes).

Neutrophils
Neutrophils may be segmented or band (immature) forms and contain cytoplasmic granules as with other mammals but may differ in cytochemistry and ultrastructure. The neutrophils of guinea pigs, hamsters, and gerbils are often referred to as heterophils or pseudoeosinophils because they contain granules that stain eosinophilic in color with Romanowsky stains. The nuclei of mature rat and mouse granulocytes may lack distinct lobes and be horseshoe, sausage, or doughnut shaped, as may those of gerbils. Rats and mice have neutrophils with colorless cytoplasm, although some dust-like red granules may be present that cause the cell to stain diffusely pink with Romanowsky stains. Cytochemical differences include hamster neutrophils that have no lysozyme activity, and decreased alkaline phosphatase (AP) activity as in the neutrophils of mice.

Eosinophils
Eosinophils make up 1% to 3% of leukocytes in rodents and contain large cytoplasmic granules that become increasingly eosinophilic as the cell matures. Eosinophils increase in number when there is a chronic antigenic stimulus such as parasite larva and allergic reactions associated with mast cells and basophilic degranulation. Eosinophils are easily distinguished from the heterophils of rodents by their larger round- to rod-shaped granules. Rodent eosinophils are typically larger than neutrophils and have a ring- or U-shaped nucleus. Their cytoplasm is often basophilic and contains small, round eosinophilic granules. In chinchillas, eosinophils are the only granulocytic cell with eosinophilic cytoplasmic granules.

Basophils
Rodent basophils make up 0% to 3% of the leukocytes and have characteristic basophilic granules on Romanowsky-stained blood films. The basophils of guinea pigs may stain reddish purple to black. Morphologically, granules of rodents are homogenous. The basophils of rodents must be differentiated from mast cells (more common when blood is taken by cardiocentesis).

Monocytes
Monocytes are the largest leukocyte in peripheral blood films and are similar in all rodents. The monocyte nuclei vary from round to oval to lobed. The cytoplasm is moderately abundant and light blue-gray in color and may contain azurophilic granules.

Lymphocytes
Lymphocytes are more variable in rodents, with variable size, cytoplasmic color (light to dark blue), and degree of nuclear chromatin condensation. Size may
vary between that of an erythrocyte and a neutrophil. Smaller cells are thought to be inactive. Reactive lymphocytes have more abundant cytoplasm that stains basophilic and nuclei with clefts. These cells are thought to be B cells involved in immunoglobulin production. The cytoplasm of true rodents stains light blue and may contain azurophilic granules in large cells. Large lymphocytes of guinea pigs often contain a single large intracytoplasmic inclusion 1 to 8 μm in diameter with finely granular to homogenous, red-staining cytoplasm, which is referred to as a Kurloff body. They are more common in young male and pregnant guinea pigs, which suggests their presence is influenced by sex hormones. These cells are thought to function as killer cells. The differential WBC count of rats tends to be lymphocytic, whereas most other rodents’ neutrophils make up a higher percentage of cells. This trend may vary with study, age, and sex of the rodent and (unfortunately) laboratory. It is best to evaluate a particular patient based on normal values for the laboratory used.

In most rodent species, the leukocytic response to disease appears to be more of an increase in immature leukocytes, toxic neutrophils, and Döhle bodies (aggregates of endoplasmic reticulum that appear as gray-blue cytoplasmic inclusions), not the total and differential leukocyte counts. Leukocytosis in rodents may be associated with chronic, closed-cavity inflammation (e.g., abscesses). Epinephrine causes neutrophils or heterophils to leave the marginating pool as a response to increased heart rate and blood flow, resulting in a neutrophilia or heterophilia. Exogenous and endogenous corticosteroids associated with physiological stress that accompanies systemic disease or is from an exogenous source will cause a lymphopenia and occasionally a mature neutrophilia/heterophilia.

**Clinical Chemistry of Rodents**

As with other domestic animals, blood chemistry values have long been investigated for common species of rodents. Changes in these chemistries are helpful in the diagnosis of many diseases. The chemistries of rodents are not different from those of the rabbit and are discussed in greater detail in the article on rabbit diagnostic testing included in this issue.

When evaluating changes in clinical chemistries, it is important to remember that these values are not only affected by alterations of metabolism and cellular dysfunction but by changes in the diet and environment as well. This may be truer for the rodent patient than any other treated by the veterinarian. Glucose values are affected by the nutritional, hormonal, and emotional state of the animal. Hyperglycemia has been reported in hamsters during hibernation. Blood urea nitrogen (BUN) levels are highly variable in rodents, affected mostly by diet. Laboratory rodent diet protein levels may vary from 12% to 24%, resulting in significant changes in BUN in healthy animals. BUN values vary widely in hamsters with nephritis, and creatinine is a more reliable indicator of renal function. Creatinine values are affected by many drugs and compounds that rodents may be exposed to, including barbiturates, glucose, protein, acetone, ketones, ascorbic acid, and sulfobromophthalein.

Unlike the rabbit, bilirubin is the end product of hemoglobin metabolism in rodents. Measurement of total bilirubin includes both conjugated and unconjugated (free) forms and is a useful screening test for liver damage or hemolysis in rodents. The AP present in the serum of a healthy hamster is composed mostly of the isoenzyme from bone and intestine. Elevated levels of AP may be found in the hamster with liver disease, but dramatic increases are usually associated with bile duct obstructions. High levels are also associated with leukemia and tumors of the prostate gland.

**Urinalysis**

Urinalysis (UA) is a simple and quick procedure that can yield valuable information regarding the function of the urinary system as well as other organs and systems in the body. In the author’s practice, it is part of the basic information collected on many rodents presented with clinical disease signs. Guinea pigs are particularly susceptible to urinary tract infections and rarely are presented with diabetes mellitus (DM). UA is performed in the rodent similar to techniques used for other mammals. Urine may be collected by free catch, manual expression, or cystocentesis. Catheterization is easily performed in larger rodent species (e.g., guinea pigs, chinchilla, prairie dogs, Degus) but is very difficult in others (e.g., mice, hamsters). When obtaining a sample by free catch, an effort should be made to avoid the first part of the urine stream because it will contain exudate flushed from the urethra. Overzealous manual expression may result in artificial blood in the urine sample. Anesthesia may be required for cystocentesis or catheterization in most patients. Urine collected for bacterial cultures should be obtained by catheterization or cystocentesis. The UA is best
analyzed as soon as the urine is collected. Urine preserved by refrigeration is suitable for examination for 2 to 3 hours.

Physical Examination of Urine
The color of normal rodent urine varies from colorless to yellowish-white to light brown. Always consider color and turbidity in association with urine-specific gravity. Rodents eating vegetables or those that have been recently treated with antibiotics may temporarily pass porphyrins in their urine, creating orange or red-tinged urine that may be confused as blood. Myoglobin or methemoglobinuria may cause rodent urine to appear dark brown. Urine from normal guinea pigs, chinchilla, and some other highly herbivorous rodents usually contains large amounts of light-colored sediment and may appear abnormal to the uninitiated. Rodents with low urine-specific gravity tend to have clearer urine containing less sediment. Cloudy urine must always be interpreted based on microscopic evaluation. The most common cause of cloudy urine is calciuria, which is normal to some extent in rodents. Other causes of cloudy urine include WBCs, RBCs, epithelial cells, bacteria, and mucous.

Specific Gravity
Refractometer measurements of urine-specific gravity are accurate and only require a single drop of urine. The author has found the urine-specific gravity measurements from the commercial urine test strips to be highly inaccurate. Normal rodent urine-specific gravity ($\rho/\rho_{H_2O}$) ranges from 1.003 to 1.050, whereas normal values for the rat range from 1.022 to 1.050. A fixed urine-specific gravity (1.008-1.012) combined with clinical dehydration and increased BUN and creatinine levels (uremia) or an abnormally high PCV is an indication that the functional competence of the kidneys is in question.

Reaction (pH)
Normal rodent urine is markedly alkaline, typical of herbivores. The pH normally ranges from 8 to 9. Low urinary pH may result from high protein diets, catabolic states, starvation, and fever. Urine stored at room temperature becomes more alkaline with time because of the decomposition of urea.

Protein
Urine protein levels may be significant in some rodents. Hamsters average about 9.7 mg/wk or about 10 times the protein excretion rate of man.

Urine Glucose
Glucose is not present in normal rodent urine; however, the author has experienced trace to even moderate amounts of glucose in the urine of otherwise healthy rodents after acutely painful or frightening experiences or with some chronically stressful situation (pain). Guinea pig patients with DM commonly present with urine glucose levels in the 300 to 600 mg/dL range.

Urine Ketone
Ketones, most often acetone, acetoacetic acid, and beta-hydroxybutyric acid, are found in rodent urine after prolonged anorexia or starvation including that associated with pregnancy. Guinea pigs with DM often present in a state of ketosis. Guinea pigs and chinchillas with severe dental disease and those that have a severely impacted cecum are the most common examples of patients that present with ketones in the author’s practice.

Urine Bilirubin
As discussed above, rodent bilirubin metabolism is similar to that of most mammals. Increased levels of bilirubin in their urine parallel increased levels in their serum. Positive urinary bilirubin is associated with the destruction of heme from blood cells or muscles or disease of the biliary tree.

Hematuria
Blood may be found in rodent urine with chemical reagent test strips or by examination of urine sediment. Test strips test positive for hemoglobin, myoglobin, and RBCs in the urine. The distinction between hematuria and hemoglobinuria or myoglobinuria requires examination of sediment and is of great diagnostic significance. Normal rodent urine has very few RBCs (0-3 cells/high-power field). An increase in RBCs occurs with inflammation or damage of the urinary or reproductive systems caused by infection, calculi, or neoplasia.

Urine Sediment
Crystaluria is common in rodents. Amorphous calcium carbonate and triple-phosphate crystals are most common in the true rodents including hamsters. Rodents taking sulfonamide drugs may have sulfur crystals. RBCs, WBCs, casts (cylinduria), and bacteria may be found in rodent urine and have the same interpretation and importance as they would in other mammalian pets. Urine collected by manual expression of the bladder may contain an abundance of RBCs. The presence of WBCs in the urine...
suggests infection. Casts suggest renal tubular disease.

Serology

The diagnosis of infections is facilitated through serologic diagnostic evaluation. Because serologic diagnostic testing measures immunoglobulins that take time and an intact immune system to develop, it may be negative at the time of clinical disease, and in immature, immunodeficient, and immunocompromised rodents. Ranges of negative and positive results for specific assays are established from control sera. There are no “normal ranges” as with serum chemistry values. Serology is the foundation for the disease-free laboratory rodent. The clinical laboratories that provide the majority of rodent serology testing primarily support research and biotechnology facilities (Table 1). Sound Diagnostics (Woodinville, WA USA) is an exception, with the majority of its clients being clinical veterinarians. The eminent rodent diagnostic laboratory is Research Animal Diagnostic Laboratory (RADIL) at the University of Missouri. RADIL offers services to veterinarians with clinical samples as will several other public and private diagnostic laboratories (Table 1).

Most laboratories use enzyme-linked immunosorbent assay (ELISA) testing to measure antibodies to the organisms associated with clinical disease in rodents. ELISA testing is relatively easy and inexpensive to perform. The amount of antibody is quantified by optical density and compared with positive and negative controls.

Some laboratories use indirect fluorescent antibody testing or Western blot testing. A new method for screening very small rodent samples for many tests used by RADIL is multiplex fluorescent immunoassay (MFI). MFI requires only 0.2 μL of undiluted serum regardless of the number of tests requested (recommended submitted sample volume to allow for potential confirmatory testing and ease of handling is 100 μL of 1:5 diluted or 20 μL of undiluted serum). The technology is based both on bead-based immunoassay and flow cytometry. Each purified RADIL antigen or control preparation is covalently linked

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**Table 1. Veterinary diagnostic laboratories that offer serology, PCR, and other services to clinical veterinarians. This list includes those laboratories known to the author and does not imply that other diagnostic laboratories do not offer these of similar services**

| Laboratory name and contact information | Testing available |
|----------------------------------------|-------------------|
| BioReliance Corporation, Laboratory Animal Diagnostic Service, (LADS), 14920 Broschart Rd, Rockville, MD 20850-3349; phone: (800)804-3586; http://www.bioreliance.com/lads_intro.html | PCR, serology, parasitology, histopathology, clinical pathology, microbiology |
| Charles River Laboratories, Inc., Research Animal Diagnostic Services, 251 Ballardvale St, Wilmington, MA 01887-1000; phone: 800-338-9680; http://www.criver.com/research_models_and_services/research_animal_diagnostics/index.html | PCR, serology, parasitology, histopathology, necropsy, microbiology |
| Division of Laboratory Animal Medicine, LSU School of Veterinary Medicine, Skip Bertman Dr, Baton Rouge, LA 70803; phone: 225-578-9643 | Serology |
| University of Miami, Comparative Pathology Laboratory, 1600 NW 10th Ave, Miami, FL 33136; phone: (800)596-7390; http://pathology.med.miami.edu/ | Serology, histopathology, necropsy, microbiology, parasitology |
| Research Animal Diagnostic Laboratory (RADIL), University of Missouri, Room W104, Veterinary Medicine Bldg, 1600 E. Rollins, Columbia, MO 65211; phone: (800)669-0825; http://www.radil.missouri.edu/ | PCR, serology, necropsy, histopathology, microbiology, clinical pathology, parasitology |
| Zoologix, Inc., 9811 Owensmouth Ave, Suite 4, Chatsworth, CA 91311; phone: (818)717-8880; http://www.zoologix.com | PCR |

*Abbreviation: PCR, polymerase chain reaction.*
| Disease                                      | Agent                      | Occurrence  | Species affected                                                                 | Clinical importance                                                                 |
|----------------------------------------------|----------------------------|-------------|----------------------------------------------------------------------------------|--------------------------------------------------------------------------------------|
| **Cilia-associated respiratory bacillus (CARB)** | Poorly classified bacteria | Common      | Mice, rats, hamsters, guinea pigs                                                | Complicates other respiratory disease                                                |
| **Clostridium piliforme (Tyzzer’s)**          | Bacteria                   | Uncommon    | Mice, rats, gerbils, hamsters, guinea pigs, rabbits, etc.                         | Clinical and subclinical infections                                                  |
| **Ectromelia virus**                          | DNA virus, family Poxvirus (Mousepox) | Rare        | Mice                                                                             | Foot swelling, pocks, lethargy, depression and sudden death, distal portions of the tail and limbs may necrose and slough |
| **Encephalitozoon cuniculi**                  | Protozoan, order Microsporidia. | Much less common than in rabbits | Rabbits, mice, rats, hamsters, guinea pigs                                       | Occasionally causes disease in rodents                                               |
| **Lymphocytic choriomeningitis virus**        | RNA virus, family Arenaviridae, genus Arenavirus | Rare        | Mice Syrian hamsters, rabbits, guinea pigs, dogs, primates, man, etc.            | Infection runting, immune-complex glomerulonephritis, death                           |
| **Kilham’s rat virus (KRV)**                  | Single-stranded DNA virus, family Paroviridae, genus Parovirus | Common      | Rats                                                                             | Subclinical infections most common. Jaundice and ataxia in young rats; clinical disease with mortality in older rats. |
| **Mammary tumor virus**                       | RNA virus, family Retrovirus, genus Betaretrovirus | Common      | Mice                                                                             | Mammary tumors                                                                       |
| **Minute virus of mice**                      | DNA virus, family Paroviridae, genus Parovirus | Common      | Wild and domestic mice                                                            | Not important clinically                                                              |
| **Mouse adenovirus**                          | DNA virus, two strains MAD-1, Mad-2 | Rare        | Mice, rats                                                                       | Mad-2 may be associated with gastrointestinal signs; Mad-1 is asymptomatic           |
| **Mouse hepatitis virus**                     | Single-stranded RNA virus, family Coronaviridae, genus Coronavirus | Common      | Mice                                                                             | Subclinical infections in adult mice, diarrhea, poor growth, and high mortality rate in neonatal mice. |
| **Mouse parvovirus**                          | Single-stranded DNA virus, family Paroviridae, genus Parovirus | Common      | Mice                                                                             | Inapparent infection, not important clinically                                       |
| **Mycoplasma pulmonis (Myco)**                | Gram-negative bacterium, family Mycoplasmamataceae | Very common | Rats, mice, wild rats, rabbits, Syrian hamsters, guinea pigs                     | Significant cause of respiratory infection in rodents                                  |
| **Pneumonia virus of mice**                   | RNA virus, Paramyxoviridae, genus Pneumovirus | More common in rats than mice          | Mice, rats, hamsters                                                            | Inapparent infection, not important clinically                                       |
| Disease                                                                 | Agent                                                                 | Occurrence   | Species affected                                                                 | Clinical importance                                                                 |
|------------------------------------------------------------------------|----------------------------------------------------------------------|--------------|----------------------------------------------------------------------------------|--------------------------------------------------------------------------------------|
| Rat parvovirus (Kilham’s rat virus, H-1)                               | Single-stranded DNA virus, family Paroviridae, genus Parovirus        | Common       | Rats                                                                             | Inapparent infection, not important clinically                                        |
| Reovirus 3                                                              | RNA virus, family Reoviridae, genus Reovirus                         | Common       | Rats, mice, wild rats, rabbits, Syrian hamsters, guinea pigs, and other rodents  | Inapparent infection, not important clinically. Stunting, diarrhea, oily coats, abdominal alopecia, and jaundice in neonatal mice |
| Rat coronavirus/sialodacryoadenitis virus—rotavirus                     | RNA virus, family Coronaviridae, genus Coronavirus                    | Common       | Rats                                                                             | Clinical signs associated with eyes, upper respiratory tract                           |
| Epizootic diarrhea of infant mice virus—rotavirus                       | RNA virus, family Reoviridae, genus Rotavirus, group A                | Not uncommon | Mice                                                                             | Diarrhea (watery, mustard-colored stool), lethargy, and distended abdomen in neonatal mice |
| Sendai virus (Sendai)                                                   | RNA virus, family Paramyxoviridae, genus Paramyxovirus                | Uncommon     | Mice, rats, hamsters, and possibly guinea pigs                                  | Respiratory disease can result with superimposed infections, e.g., *Mycoplasma pulmonis*, CAR bacillus |
| Theiler’s murine encephalomyelitis virus                                | RNA virus, family Picornaviridae, genus Enterovirus                   | Not uncommon | Mice, rats                                                                       | Central nervous system disease (paralysis, seizures, vestibular, etc.)                |
| Toolan’s H-1 virus (H-1)                                                | Single-stranded DNA virus, family Paroviridae, genus Parovirus        | Uncommon     | Rats                                                                             | Inapparent infection, not important clinically                                        |
to 1 of 100 different types of polystyrene beads, which vary slightly in the intensity of their color. If immunoglobulin G antibody to a particular antigen is present, it will bind to the antigen on a specific bead and will then be detected by subsequent binding of goat antimouse antibody conjugated to a fluorochrome, R-phycoerythrin. The reader channels single beads through a dual-laser detector, which simultaneously determines both the bead type by the internal dye combination and the fluorescent intensity associated with each individual bead. The fluorescent intensity associated with each of 100 individual beads of each type is used in the determination of each MFI value. The overall correlation between MFI and ELISA is greater than 99.5% for both mouse and rat samples. In general, MFI is more sensitive than ELISA and is less prone to false-positive results.

**Polymerase Chain Reaction**

Polymerase chain reaction testing is now available to the clinical veterinarian treating rodent patients. Depending on the primers chosen, polymerase chain reaction testing can be a very specific and sensitive test. One laboratory that provides services to both research/biotech and clinical veterinarians offers a set of 8 tests run on a single mouse fecal pellet (Zoologix, Inc., Chatsworth, CA USA).

**Rodent Infections**

Table 2 lists many of the diseases for which serological testing is available. Some of these diseases are important only because of their influence on scientific research or other bioscience.

### References

1. Bannerman R: Hematology, in Foster H, Small D, Fox JG (eds): The Mouse in Biomedical Research, Volume III. New York, Academic Press, pp 293-312, 1983
2. McGuill MW, Rowan AN: Biological effects of blood loss: implications for sampling volumes and techniques. *ILAR News* 31:5-20, 1989
3. Sakaki, K: Hematological comparison of the mouse blood taken from the eye and the tail. *Exp Anim* 10:14-19, 1961
4. Hoff J: Methods of blood collection in the mouse. *Lab Animal* 29(10):47-53, 2000
5. Benjamin MM: Outline of Veterinary Clinical Pathology (ed 3). Ames, IA, Iowa State Press, 1978
6. Probs RJ, Lim JM, Bird DN, et al: Gender differences in the blood volume of conscious Sprague-Dawley rats. *J Am Assoc Lab Anim Sci* 45(2):49-52, 2006
7. Sisk DB: Physiology, in Wagner JE, Manning PJ (ed): The Biology of the Guinea Pig. New York, Academic Press, pp 63-98, 1976
8. Ansari A, Williams JF: The eosinophilic response of the rat to infection with *Taenia taeniaeformis*. *J Parasitol* 62(5):728-736, 1976
9. Campbell TW, Ellis C: Avian and Exotic Animal Hematology and Cytology (ed 3), Oxford, Blackwell Publishing, 2007
10. Tomson FN, Wardrop KJ: Clinical chemistries and hematology, in *Van Hoosier GL, McPherson CW* (eds): *Laboratory Hamsters*. Orlando, Academic Press, pp 43-58, 1987
11. Ringler DH, Fabich L: Hematology and clinical biochemistry, in *Baker HJ, Lindsey JR, Weisbroth SH* (eds): *The Laboratory Rat*, Volume I. Orlando, Academic Press, pp 105-121, 1979
12. Tietz NW: Fundamentals of Clinical Chemistry, Philadelphia, Saunders, 1976