The exotic small mammal physical examination is performed similarly to exams of other small animals such as dogs and cats. All items that may be needed during the physical examination should be out and within hand’s reach before you start. Exotic small mammals can become easily stressed, so it is essential to keep the “time in hand” to a minimum when necessary. Before you start the hands-on portion of your examination, perform a visual examination. The visual examination should include information about the cage as well as the appearance and mentation of the animal. It is important to note any toys, furniture, and bedding in the cage as well as the diet offered to the animal.

Obtaining the patient’s weight and temperature should be the first part of the physical examination. Since many of the small mammal patients are very small, it is best to use a scale that weighs to the nearest gram. Obtaining a temperature can be challenging, but a digital thermometer is generally the quickest and best method.

A general rule of thumb for performing a physical examination is to start at the head and work your way down to the tail. This will help ensure nothing is overlooked. As stated above, the weight and temperature should generally be obtained first. It is also a good idea to obtain both a heart and respiratory rate as soon as the animal is removed from the cage. If the respiratory rate seems exaggerated, recount the rate upon completion of the examination and after the animal has been placed back into the cage. The primary deviation from the “head to tail” method of a physical examination in small mammals is the oral examination. In dogs and cats the mouth is usually examined when the head is examined. The oral examination can be very stressful for small mammals (ferrets are the exception) and therefore is generally done last.1

Continue the examination by looking at the eyes, ears, and nares. They should be clean, clear, and free of any discharge. Any signs of discharge or debris from the eyes, ears, and/or nares can be a sign of an underlying disease process and should be investigated further.

It is important to palpate the limbs and the abdomen during your physical examination. Palpation should include checking for any masses (especially in rats), old wounds or lesions, or any other potential abnormalities on the body. In many species of small mammals, the stomach, kidneys, cecum (if present), spleen (especially in ferrets), and bladder (if full) can be easily palpated. After palpating the abdominal cavity, examine the feet and genitalia. It is also important to examine the skin and look for any signs of dermatophytes or ectoparasites.

The heart and lungs should be ausculted using an infant or pediatric stethoscope. It is important to note the presence of any potential heart murmurs, arrhythmias, harsh lung sounds, or anything that may sound abnormal. This is performed in the same manner as would be performed on a dog or cat. A body condition score should also be assigned to the patient. The same system used with dogs and cats is also used with small mammals, with 1 out of 9 being emaciated and 9 out of 9 being grossly obese.

The hydration status of the patient should be evaluated during the physical examination. This is done similarly to a dog or cat. The mucous membranes should be moist and pink. As with most other mammals, the capillary refill time should be between 1 and 2 seconds. A common sign of dehydration is dry or tacky mucous membranes. Another sign of dehydration is sunken eyes and lack of skin turgor. The skin should be tented or pulled upward to assess dehydration.

The mouth is usually examined last. The oral examination can be difficult in an awake patient; therefore light sedation or general anesthesia may be required on patients that are not cooperative. Common drugs used for sedation include midazolam and butorphanol. Isoflurane or sevoflurane are the two inhalant anesthetics used to provide general anesthesia.1, 2 Ferrets are an exception. The ferret oral exam can be performed when the head is being examined (very similar to a dog or cat oral examination).

A mouth speculum with a light source is one of the most helpful instruments that can be used when performing an oral examination (for rodents and rabbits only). There are a few different mouth specula that can be used. A long otoscope cone attached to the otoscope handle can be used to examine the mouth.3 A small vaginal speculum and a pen light can also be used. A rigid endoscope along with a mouth speculum can be used to examine the mouth.8
of the most effective ways to perform an oral examination on a small mammal is with a bivalve nasal speculum. This instrument (manufactured by Welch Allyn®*) has a light source and attaches to a Welch Allyn® battery hand piece. It is important to examine the gums, tongue, and all of the teeth, including the incisors. Look for any dental abnormalities such as malocclusion, tongue entrapment (generally seen in guinea pigs with severe dental disease), incisor overgrowth, fractured teeth, or points on the lingual or buccal surfaces of the premolars and molars (common in guinea pigs, rabbits, and chinchillas). If dental disease is present, a further work-up may be necessary, including bloodwork, radiographs, etc.

Once a complete physical examination has been performed and the animal is stable, diagnostic tests such as a fecal, bloodwork, radiographs, etc. (if needed) should be done.

**Ferret Capture and Restraint**
Ferrets are restrained similarly to cats. Most of the examination can be performed with a little petting and treats such as a high calorie paste. When more restraint is necessary, the ferret can be restrained like a cat.

**Rabbit Capture and Restraint**
Rabbits can usually be picked up out of their enclosure and placed on the exam table. It is important to note that with improper restraint, rabbits can kick out their hind legs and fracture their backs. Many cooperative rabbits can be lightly restrained by petting them and gently holding them, making sure they do not jump off the table. If more restraint is necessary, the rabbit can be restrained by tucking the head between the side of your body and your arm. The other arm should support the rest of the rabbit’s body against your own body. Essentially it looks like you are tucking a football against your body.

**Chinchilla Capture and Restraint**
Most pet chinchillas are used to being handled and are therefore generally easy to capture and restrain. When catching a chinchilla, it is best to place one hand under the thorax/abdomen and with the other hand gently grasp the base of the tail. Once the chinchilla has been captured, it can be placed on the exam table. When a chinchilla becomes frightened, it can lose patches of fur if handled roughly. This is commonly referred to as “fur slip.” Due to “fur slip,” a chinchilla should generally not be scruffed. Although not very common, “fur-slip” can happen unexpectedly during the physical examination. It is therefore a good idea to warn the client in advance that this possibility exists. If the client is educated in advance about “fur-slip,” she will generally be much more understanding if it happens.

**Rat Capture and Restraint**
Most pet rats are used to being handled and can simply be picked up and gently supported during a physical examination. If more restraint is necessary, the rat’s head can be restrained by placing your thumb on one side of the head and your index finger on the other side while still supporting the body. The rat can also be gently scruffed if necessary. For rats that are uncooperative, the base of the tail can be gently grabbed to temporarily catch the patient before restraining it. This is generally only used if the animal is not tame or used to being handled. This can also be used when handling mice.

**Hamster and Mouse Capture and Restraint**
Hamsters have a large amount of loose skin around the neck, shoulders, and back. To provide full or immobilizing restraint, the hamster’s skin should be grasped between your thumb and fingertips (similar to scruffing a cat). It is important to always support the hamster’s body with your other hand. Mice are restrained in a very similar manner, but can be picked up by gently grabbing the base of the tail before scruffing them.

**Guinea Pig Capture and Restraint**
Most pet guinea pigs are calm and gentle animals that rarely bite; therefore they can usually be picked up out of their enclosures and placed on an exam table. Under many circumstances, guinea pigs will need only light restraint when being examined. This is usually best accomplished by supporting the hind end of the animal with one hand and cupping the other hand gently under the thorax. Scruffing a guinea pig is generally not recommended because the neck is very short and there is little skin to easily grasp and grip.
**Ferret Venipuncture Sites**

Common venipuncture sites include the cephalic, lateral saphenous, jugular, and cranial vena cava vessels. The peripheral vessels are small, which makes it sometimes difficult to obtain a full panel. Blood can be obtained from the jugular vein in a similar manner as in a dog or cat (note that the skin is thick and the vessel is more lateral than on a dog or cat). The cranial vena cava is generally the quickest method that may yield the largest amount of blood. If the cranial vena cava is used, the animal must be placed under general anesthesia before the sample is obtained. A 25-gauge needle attached to a 1 or 3 cc syringe is used for this venipuncture site. The patient is placed in dorsal recumbency with the front legs pulled down next to the body and the head and neck extended. The needle should be aimed toward the opposite hind limb and is then inserted at a 45 degree angle into the thoracic inlet. The landmarks used to find the insertion site include the manubrium and the first rib. Do not poke and jab this vessel trying to obtain a sample, as it could be dangerous for the patient. This is the same technique used in guinea pigs.

**Rabbit Venipuncture Sites**

Venipuncture sites include the cephalic, lateral saphenous, auricular, and jugular veins. The jugular veins can be difficult to obtain blood from in rabbits that have large dewlaps. Performing jugular venipuncture on rabbits is done in the same manner as described for chinchillas. The cephalic and auricular vessels are generally small and may not yield enough blood for a full blood panel. The lateral saphenous vein is usually the quickest and easiest vein to obtain a blood sample from. Obtaining blood from the lateral saphenous in a rabbit is accomplished in the same manner as in a dog. A 1 mL syringe with a 27- or 25-gauge needle should be used. Proper restraint is a must.

**Chinchilla Venipuncture Sites**

Venipuncture sites in the chinchilla include the cephalic, lateral saphenous, and jugular veins. The peripheral vessels do not usually yield enough blood for diagnostic sampling; therefore the jugular vein is generally the vessel of choice when obtaining blood for a complete blood count and a biochemistry panel. Performing jugular venipuncture on a chinchilla is similar as on a cat (be aware of “fur-slip”). The chinchilla can be held in one of many different positions to gain access to the jugular vein. Choosing the position will depend on the animal as well as what preference the phlebotomist has. The chinchilla can be placed in lateral recumbency with the head extended and the legs pulled down toward the body. Another position involves the patient being placed in sternal recumbency at the edge of the exam table. Once properly restrained, the head can be extended upward and the front legs pulled down (also similar to a cat). The last common position includes the chinchilla being rolled onto its back. The head is extended and the legs are pulled down toward the body. The vessel can then be held off using your thumb or index finger (same as in a cat). The jugular vein is located in the same general area of the neck as the jugular vein in a cat. A 27- or 25-gauge needle attached to a 1 mL syringe is commonly used to obtain the blood sample. Animals that are uncooperative may need light sedation to obtain the sample.

**Guinea Pig Venipuncture Sites**

The peripheral vessels as well as the jugular vein can be used to obtain a blood sample from guinea pigs. The needle and syringe size as well as the positioning are the same as for chinchillas. Again, the peripheral vessels may not yield enough blood if a complete blood count and biochemistry are needed. Guinea pigs have very short necks; therefore the jugular vein can be difficult to draw blood from. In the author’s opinion, the femoral vein is the “better” vessel to obtain a blood sample from. Obtaining blood from the femoral vein is generally the quickest method that yields the largest amount of blood. The sample can be taken once the patient is properly anesthetized. The patient is anesthetized to minimize the chance of lacerating the large femoral vessel. To obtain the blood sample, first palpate the pulse of the femoral artery in the inguinal area. The nipple can be used as a landmark to help find the pulse. Because the artery and the vein run alongside each other, the arterial pulse is used to help pinpoint the location of the vein. Once the artery is palpated, the needle (it is best to use a 1cc tuberculin syringe with a 25- or 22-gauge needle) should be inserted at a 45 degree angle into the skin. This venipuncture site is a “blind stick.” Since the vessel cannot be visualized, it is important to place slight negative pressure on the syringe as soon as the needle enters the skin. Having negative pressure on the syringe is helpful for the phlebotomist because blood will enter the syringe once the needle is properly placed into the vessel. Without negative pressure on the syringe, it is very easy to bypass the vessel and not even realize it. Once the blood sample has been obtained, the needle can be removed and the thumb or a finger placed over the insertion site to hold off the vessel.

Blood can also be obtained from the cranial vena cava (same procedure as used in a ferret), but this does not come without potential risk of traumatic bleeding into the thoracic cavity or pericardial sac. If the cranial vena cava is chosen as the venipuncture site, the animal must be under general anesthesia while obtaining the blood sample. A
struggling patient can greatly increase the risk of lacerating the vena cava during the blood draw; therefore this
venipuncture site should never be used when an animal is awake.

**Rat, Mouse, and Hamster Venipuncture Sites**
The peripheral veins as well as the jugular vein can be used, but again, these vessels are small and are hard to obtain
a diagnostic sample from. In rats and mice, the lateral tail veins can be used to obtain small amounts of blood. The
vessels are usually superficial and can easily be seen (unless the animal is very debilitated or obese). This can be
accomplished by using either an insulin syringe or tuberculin syringe with a 27-gauge needle. A 27- or 25-gauge
needle can also be placed into the vessel. Once there is blood in the hub of the needle, a hematocrit tube can be used
to collect the blood. Generally, this site does not yield enough blood for a complete blood count and biochemistry.
Using the femoral vein to obtain a blood sample is generally the quickest method that will yield the most amount of
blood. The same method described in guinea pigs is also used with hamsters, mice, and rats, although a 27- or 25-
gauge needle is generally used.4

**References**

techniques of guinea pigs and chinchillas. In *Ferrets, rabbits, and rodents: Clinical medicine and
Saunders.
Saunders.
298. St. Louis: Saunders.
Lake Worth, FL: Zoological Education Network.