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Rhinoceroses

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The Rhinocerotidae

Introduction

Like the fabricated creature in Albretch Dürer's famous lithograph, the rhinoceros has long been a source of mystery, myth, and intrigue. Part unicorn and part armored beast, the current knowledge of rhinoceros anesthesia likewise represents a melding of pure art and hard science. Today rhinoceros anesthesia is relatively commonplace, yet no less demanding in practice.

The Rhinocerotidae are truly living fossils – a remnant and archaic mammalian family represented by only five extant species in four genera restricted to Africa and Asia. The relic survivors belie a one-time place of dominance among vertebrate organisms with over 150 fossil rhinoceros species discovered by paleontology across four continents (Prothero 2005). Yet, only five rhinoceros species are extant, four of which are critically endangered from poaching and loss of habitat, while the fifth is under constant relentless poaching pressure with current population numbers scarcely sustaining losses, and only through great resource investment from conservationists (Foose & van Strien 1997; Emslie & Brooks 1999).

Field anesthesia made possible the rhinoceros conservation success stories of the twentieth century (Harthoorn & Lock 1960; Player 1972; Meadows 1996) and remains a critical tool for proactive rhinoceros management programs incorporating translocation, ear-notching, radiotelemetry, microchip implantation, proactive horn trimming, and other techniques designed to secure the conservation of both African and Asian species (Dinerstein et al. 1990; Dinerstein 2003; Kock et al. 1990, 1995). Historical rhinoceros anesthesia protocols were based on highly effective reversible opioid combinations, yet new anesthesia techniques continue to improve efficacy and safety for both animals and human personnel (Fig. 47.1).

Taxonomy and Evolutionary History

The odd-toed ungulates of the order Perissodactyla include three living families: the rhinoceroses, horses, and tapirs. As the order name denotes, all Perissodactylids bear weight on one (equids) or three (rhinoceroses and tapirs) digits. Rhinoceroses and tapirs are among the most primitive of the world's large mammals and are further grouped into the suborder, Ceratomorpha based on a similar ancient body plan. The stout body of the rhinoceros is graviportal or designed for weight bearing with limb modifications to support large mass rather than the long angular limbs of equids in the suborder, Hippomorpha, specialized for speed.

The Perissodactyla enjoyed a period of extraordinary diversity in the Eocene epoch 34 to 55 million years ago before climate change presumably limited species radiation, culminating in extinction of 10 of the 14 perissodactyl families by the end of the Oligocene (Radinsky 1969). Prehistoric rhinoceroses in particular, as interpreted from fossil evidence, represented a far bigger group of organisms than exist today and included both horned and hornless forms. In fact, rhinoceroses were once the most common large herbivore in North America for most of the last 50 million years (Prothero & Schoch 2002). An extinct hornless rhinoceros, named Paraceratherium (also called Indricotherium), is known to science as the largest land mammal that ever lived, measuring over 6 m at the shoulder and weighing an estimated 20 tons (Prothero & Schoch 2002; Prothero 2005).

Biology and Morphology

The Rhinocerotidae are large terrestrial herbivores that have evolved either a browsing (black, Sumatran, Javan) or grazing (white, greater one-horned) strategy to process large quantities of fibrous feeds or simple grasses, respectively. As such, they share bulky elongated skulls, dental

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Figure 47.1 Helicopter-assisted aerial slinging of a black rhinoceros (*Diceros bicornis*) by its feet during translocation operations in the Kaokoveld of northwestern Namibia.

patterns largely devoid of canines and incisors (retained to various degree in the Asian species), and prehensile or wide, flat lips in the browsers and grazers, respectively. Like the equids, fermentation takes place in the cecum and colon. The rhinoceros gut is less efficient than that of ruminants since the microfloral protein formed in the hindgut is largely unavailable to the animal. As a result, rhinoceroses must eat more, have a relatively fast passage of gut contents, and possess limited time to reabsorb water from the feces. Therefore, rhinoceroses must drink every day or every second day, making it a water-dependent species rarely found more than 15 km from a water source.

Despite their often-conspicuous absence in many fossil rhinoceroses, the single horn (*Rhinoceros* sp.) or pair of horns (*Ceratotherium*, *Diceros* and *Dicerorhinus* sp.) is certainly the most distinguishing feature of the living Rhinocerotidae, giving name to the group literally as the *nose-horned beasts* (Prothero & Schoch 2002). Rhinoceros horns differ from true horns of the Artiodactyla by having no central core of bone. Instead, the tubular hair-like keratin filaments are

Figure 47.2 The distinctive hair coat of a Sumatran rhinoceros, a feature linking the primitive *Dicerorhinus* genus with its prehistoric past. Image courtesy of Mohd Khan bin Momin Khan, Malaysia Department of Wildlife and National Parks.

compressed in a linear fashion and set on a bony protuberance of the skull (Raubenheimer et al. 2015). Underneath the horns, the skull incorporates extensive nasal bones and sinuses – structures inordinately prone to complications from trauma during capture and translocation.

Rhinoceros skin is thick, with several centimeters of primarily collagenous dermis, with the Asian species having subdermal plates and heavy skin folds (Cave & Allbrook 1958; Shadwick et al. 1992). The greater one-horned rhinoceros of Asia is perhaps best known for the exaggerated armour-like plates or folds first popularized in the famous Dürer woodcutting of the Middle Ages. The epidermis, however, is very thin (1 mm) and heavily keratinized, incorporating extensive vasculature, which may predispose the rhinoceros to pressure necrosis, particularly in calves (Cave & Allbrook 1958; Gandolf et al. 2006). Significant body hair is an antiquated trait retained in all but one living species, the Sumatran or "hairy rhinoceros" so-called for its shaggy coat of hair (Fig. 47.2). Wild *Dicerorhinus* have shorter more bristly coats than their captive relatives, a trait providing protection for the skin from the numerous biting insects that share its environment. Hair, a primordial trait of many fossil rhinoceroses, including the woolly rhinoceros *Coleodonta* and massive one-horned hairy *Elasmotherium*, eloquently links the Sumatran rhinoceros with its long and prosperous past.

Rhinoceros Immobilization and Capture

Rhinoceros Capture Beginnings

Before widespread application of chemical capture techniques, early African rhinoceros capture operations utilized ropes and a chase vehicle (made famous in the film

Hatari staring John Wayne and Hardy Kruger). Although dangerous to the operator and stressful to the animal, some teams in East Africa became remarkably proficient at this form of capture. Chemical capture of rhinoceroses was first attempted with the dissociative anesthetic phencyclidine and the curariform muscle relaxant gallamine triethiodide. In 1960, during Operation Noah, many black rhinoceroses (*Diceros bicornis*) were saved from the rising waters of the newly constructed Lake Kariba, bordering Zambia and Zimbabwe, using these novel techniques (Child & Fothergill 1962; Condy 1964; Harthoorn & Lock 1960; Meadows 1996). Phencyclidine and gallamine were succeeded by the easily reversible opioids, first morphine and diethylthiambutene followed quickly by the more potent opioids including etorphine HCl (M99) (King & Carter 1965; King 1969; Keep et al. 1969). Over the past 60 years, etorphine has become the standard opioid for capture of the African and Asian rhinoceroses, with fentanyl citrate (Sublimaze), carfentanil citrate (Wildnil), and thiafentanil oxalate (Thianil) proving useful alternatives. Pioneering investigation by early practitioners such as Toni Harthoorn, Eddie Young, Ian Hofmeyr, Ian Player, and many others provided the foundation on which future rhinoceros chemical capture methods, including the present work, are based (Player 1972; Harthoorn 1973; Young 1973).

Remote Drug Delivery: Equipment and Darting Techniques

An assortment of remote drug delivery equipment is available for rhinoceros capture, including new developments, yet some of the early systems are still in common practice today, attesting to their simple and dependable design. In captive and boma situations all darting systems can be utilized, but nylon darts (Daninject or Telinject with 60×2 mm smooth needles) are preferred as they are quiet and relatively atraumatic. The authors prefer to hand-inject (using appropriate human protective safety measures) or pole-syringe *captive* rhinoceroses, including animals held in bomas, to eliminate the excitability that typically follows from the noise and impact of projectile darting.

For field capture of rhinoceroses on the ground or from a helicopter a robust and reliable darting method such as the Cap-Chur system is preferable, although it is a matter of personal preference, with many operators also using DanInject® or disposable Pneudart®/Motsumi® darts. Dart barrels made of aluminum or stainless steel are the most durable for field use, especially since power settings and impact energy are high, wind or down drafts from the helicopter can be significant, and the operator is often forced to shoot through vegetation. The dart needle should be 5–6 cm long for adult rhinoceroses. Rhinoceros skin can plug the lumen of a dart needle unless the needle has a relatively thick wall and narrow lumen (Cap-Chur NCL needles) or the tip is bent over (Fauncap dart needles) or the point is sealed and side ports are provided. The needle must preferably have a bead, low barb, or small collar about 25 mm from the base to hold the dart in the thick skin, although many operators successfully use 60-mm smooth needles in field conditions. A novel spiral-threaded needle was developed by Deon Joubert specifically for use on thick-skinned pachyderms; the dart needle can be easily removed by screwing it out of the animal, thereby reducing skin and tissue trauma (Fig. 47.3; Joubert Capture Equipment, Hadison Park, Kimberley, South Africa).

Proper dart placement is essential to ensure good drug deposition. The dart should be placed perpendicular to the skin for deep intramuscular (IM) injection (the thick skin of a rhinoceros often makes an angled shot ineffective). When darting from the helicopter, the muscles of the rump or the upper part of the hind leg offer the best targets. In the boma or on foot, any large muscle mass can be used for dart placement, although the neck and shoulder are

Figure 47.3 Robust dart needles for the capture of free-ranging rhinoceroses demonstrating the various design configurations of needle tip and barb. The spiral-threaded needles on the right are less traumatic and can be screwed back out of thick rhinoceros skin. Photograph courtesy of Joubert Capture Equipment, South Africa.

preferable. To avoid bounces, a dart should not hit too hard; some practitioners even say that the dart must land on the rhinoceros (C. Moueix, pers. comm. 2019). Antibiotics are routinely infused into dart wound sites, and intramammary mastitis preparations work well for this application.

Recumbency and Positioning

Recumbency and positioning are critical considerations for safe anesthesia of rhinoceroses whether in a zoological setting or in the wild. Prior to induction in captivity, thick padding or heavy mats should be utilized to protect recumbent animals from the concrete floors common in these environments. Myositis and neuropathy are serious potential complications. Traditionally, rhinoceroses immobilized in the field were maintained in or moved into sternal recumbency, however, irreversible muscle damage has developed in this position (especially if the rhinoceros goes down on a slope facing upwards with the full weight on its hind legs) as a result of occlusion of the blood supply to the limbs. Although uncommon, problems even occur with careful "placement" of the legs in an apparently natural position. With the rhinoceros in lateral recumbency, blood flow to the limbs is improved and circulation to the muscles allows delivery of oxygen and dissipation of carbon dioxide and heat generated while running. With the animal in lateral recumbency, the legs should be physically "pumped" up and down by hand every 20 minutes to aid circulation. The weight must be taken off the lower legs to "pump" them effectively; this is accomplished by two or more people lifting the upper legs and rolling the animal partially onto its back about 45° from horizontal. We recommend that all black rhinoceroses that have undergone any degree of exertion be placed in lateral recumbency for at least a few minutes.

The decision to move white rhinoceroses onto their sides should be based on several factors, including the degree of exertion, presence of muscle tremors, and duration of recumbency. White rhinoceroses often experience significant muscle rigidity, paddling, and even convulsions under opioid anesthesia; these effects are exacerbated by lateral positioning but tend to resolve with time. Importantly, the paddling and rigidity can be controlled by the post-induction administration of butorphanol, which improves blood gases by reducing the tremors and related oxygen consumption of muscles in etorphine-immobilized white rhinoceroses (Buss et al. 2018). Therefore, white rhinoceroses should be positioned initially in sternal recumbency until complete relaxation is achieved (Kock et al. 1995).

The position in which a rhinoceros is placed during recumbency is also an important consideration for respiratory function. In black rhinoceroses, it has been shown that arterial oxygen hemoglobin saturation (SaO_2) and partial pressures (PaO_2) are higher in sternal than lateral posture. Consistent with observations in domestic animals and humans, the black rhinoceros has greater dead space in lateral compared to sternal posture as indicated by a lower end-tidal carbon dioxide and higher dead space ratios and volumes in lateral (Morkel et al. 2010). The most appropriate posture for rhinoceroses during anesthesia must be based on the circumstances of each capture and should strike a balance between maintaining respiratory function while providing optimal circulation to the limbs. An oblique dorsal recumbency is probably ideal, with regular turning onto the opposite side every 20–30 minutes and a few minutes before reversal. This may require quite a few assistants in large adult rhino and provision must be made for about 10 strong assistants for longer procedures.

Eyes and Ears

The eyes of the recumbent rhinoceros must be shielded with a large towel or appropriate-sized blindfold to prevent retinal damage from direct sunlight, dirt accumulation, and corneal abrasion from the environment. For black rhinoceroses undergoing translocation, the use of a muslin cloth wrapped tightly around the face several times and secured to the horn with cable ties is preferred to a simple blindfold (Fig. 47.4). This technique serves to fix the eyes in a closed position and quiets the rhinoceros during crate transport. Before blindfolding, foreign material should be washed from the eyes using physiologic saline. The ear canals are plugged with cotton wool or a cloth while the

Figure 47.4 Attachment of a muslin cloth blindfold to aid in crate transport of the black rhinoceros (*Diceros bicornis*). Note the use of cable ties in front of the caudal horn to secure the blindfold over both eyes.

rhinoceros is anesthetized, leaving tabs for quick removal. Alternatively, when a large number of rhinoceroses are to be immobilized, two cloth-covered cotton wool plugs can be joined with cord so they remain together. If the rhinoceros is being transported, its ears are normally blocked for the trip. However, in white rhinos or animals that are pushing, it is better to remove the ear plugs since a stimulated rhino is less inclined to push than one protected from external stimuli. Caution should be used on removal of the earplugs as the sudden auditory stimulation can result in excitement or aggression.

Anesthesia Monitoring

A one-off thorough clinical examination with regular monitoring of vital functions (respiration, temperature, heart rate, capillary refill time) must be conducted for the duration of anesthesia. The focus should be on monitoring respiratory and cardiovascular function as well as thermoregulation since hypoxia and hyperthermia are common life-threatening sequelae. These functions are very much dependant on the degree of exertion and excitement before and during induction and must be kept in mind during the evaluation. Careful monitoring is especially important in old, debilitated, very young, and heavily pregnant animals or animals that become recumbent in a shorter than normal time (less than 5–6 minutes after darting). Any residual dart contents should be checked to see if they were injected, especially if more than one dart was used, as the success of drug delivery may dictate protocols for anesthetic monitoring and antidote administration.

Pulse oximetry (SpO_2, peripheral oxygen saturation of hemoglobin) provides an indirect measure of the arterial oxygen saturation of hemoglobin (SaO_2) and is a valuable aid to help monitor blood oxygenation and pulse in anesthetized rhinoceroses. However, it should not be a replacement for thorough patient monitoring. Without simultaneous correlation with arterial blood gases, pulse oximetry is a tool best used to monitor trends in oxygen saturation. SpO_2 values often fluctuate so the pulse oximeter should be observed for at least 1 minute; if the SpO_2 does not consistently read above 80% or is declining, an intervention is warranted. Based on higher oxygen affinity of white rhinoceros hemoglobin, it has been suggested that SaO_2 levels (calculated from PaO_2 by blood-gas devices and ranging from lows of 40% up to 98%; Kock et al. 1995; Atkinson et al. 2002) in rhinoceroses underestimate the true oxygen saturation of hemoglobin when calculations are made using human formulae (Bush et al. 2004; Haymerle et al. 2016). Historically, the sensor clip was attached to the pinnae of the ear after careful scraping with a sharp blade to remove the epidermis, but this technique has been linked to falsely elevated oxygen saturation of hemoglobin (SpO_2 90–100%) in hypoxemic animals (PaO_2 below 30 mmHg), presumably from measurement of blood that is exposed to air (Meyer, pers. comm., 2020). The use of a transflectance probe under the third eyelid or on the mucosa of the oral cavity, penis, vulva, or rectum is a better alternative. A cloth is placed over the sensor as ambient light can affect the reading. In animals with excessive muscle rigidity or tremors, as is common in immobilized white rhinoceroses, the sensor may fail to obtain an accurate reading. A reflectance probe held against the nasal mucosa works well (applied beyond the pigmented area) and has also been used with varying success on the inner surface of the lips, against the gums, on the conjuctiva and in the rectum or vagina. With experience, the color of the blood can be used as a crude indicator of blood oxygenation in the absence of sophisticated equipment, especially arterial blood collected from the inner surface of the pinnae.

Respiratory Gases: Oxygen and Carbon Dioxide

Respiratory depression is the most significant life-threatening complication encountered during routine anesthesia of the rhinoceros (Heard et al. 1992; Kock et al. 1995; Atkinson et al. 2002; Bush et al. 2004, 2005; Fahlman et al. 2004). Rhinos experience respiratory depression and perfusion-ventilation inequalities because of large body size, positioning, muscle rigidity, abdominal organs pressing on the diaphragm, and sensitivity to the respiratory depressant effect of strong opioids, more specifically the μ-opioid receptors (Bush et al. 2011). Severe respiratory compromise with hypoxemia, hypercapnia, and acidosis is more common with long captive procedures or under field conditions where higher doses of opioids are used to shorten induction times (Heard et al. 1992; Kock et al. 1995). Among the African species, these physiologic changes are more prevalent in the white than black rhinoceros (Bush et al. 2004, 2005).

Respiration is the first and most critical function to be monitored in rhinoceroses under anesthesia. In the field situation it is valuable to have a reliable person who does nothing but watch respiration, noting the rate and depth by observing chest movement. Be sure there is a free flow of air in and out of both nostrils and that the blindfold does not restrict airflow. The lower nostril, in particular, should remain clear of obstruction so passive regurgitation of stomach contents is free to drain. A cloth sack can be placed under the head to limit inhalation of dust or other debris. When monitoring breaths on a bouncing vehicle as with an immobilized rhinoceros transported on a sledge where it is difficult to watch chest movement, a finger can be hooked in the nostril or a hand held close to the nares to feel for the warm exhaled air. Breathing should be deep

and regular. Respiratory rate is approximately 10–15 breaths per minute on induction, going down to 4–8 breaths per minute about 10 minutes post induction when using potent opioids. Respiration must be monitored for at least 30–60 seconds to obtain an accurate picture of ventilatory pattern as immobilized rhinoceroses often give two or three quick breaths followed by a period of apnea. Rhinoceroses often develop apnea when moved into a different position. Animals should be rolled slowly and watched for breathing. A painful stimulus often incites the apneic rhinoceros to take a breath. Observation of arterial blood color by sample collection from arteries located on the inner pinna of the ear provides a reliable early indicator of blood oxygenation. Dark red, almost black blood indicates poor oxygenation while a lighter red color is normal and correlates well with mucous membrane color.

If breathing is slow, the rhinoceros develops apnea, or blood oxygenation is poor (SpO_2 consistently less than 80%), partial reversal with nalbuphine (Nubain), diprenorphine (M50:50), or butorphanol (Torbugesic) or complete reversal with naltrexone HCl (Trexonil) are indicated. In white rhinoceroses, partial agonist-antagonists are given immediately on arrival at the immobilized animal. Nalorphine is no longer available in Africa so its use has largely been replaced by alternative partial agonist-antagonists (Tables 47.1 and 47.2). Butorphanol administered intravenously at 10–20× the etorphine dosage in milligrams produces similar effects to Nalorphine (Haw et al. 2014). Since black rhinoceroses stand up readily with very small volumes of partial agonists, caution should be exercised to avoid sudden arousal. Intravenous (IV) doxapram HCl (Dopram; black rhinoceros 200 mg, white rhinoceros 400 mg) provides a smaller, transient improvement in respiratory rate and depth, and is a good "kick-starter" if apnea occurs. Doxapram must be used with caution in white rhinoceroses as it causes central nervous system excitation and exacerbates muscle tremors; it is advisable to use Doxapram in conjunction with butorphanol and supplemental oxygen.

Nasal or tracheal insufflation of oxygen (O_2; 15–30 L/min) can produce a rapid and significant increase in blood oxygen saturation in an immobilized rhinoceros but must only be administered after butorphanol administration (Haw et al. 2014, 2015). Although it did not correct systemic acidosis or hypercapnia, O_2 insufflation did substantially improve oxygenation and anesthetic safety in field-immobilized rhinoceroses (Bush et al. 2004; Fahlman et al. 2004; Haw et al. 2014, 2015). A variety of factors influence pulmonary blood gas exchange, including dose of anesthetic drug, position of the rhinoceros during immobilization, size of the animal, pulmonary perfusion and pressures, and ventilation-perfusion matching. Oxygen supplementation at the flow rates commonly used for rhinoceroses appears to produce a more profound improvement of patient oxygenation (PaO_2 108–194 mmHg) in subadult African rhinoceroses compared to adults, perhaps indicating greater ventilation-perfusion mismatch with larger body size (Fahlman et al. 2004).

A control valve and flow meter are attached to the O_2 bottle and oxygen is administered via a flexible silicon or rubber nasogastric tube (edges rounded to prevent damage to nasal mucosa), measuring 2 m long and with 9–14 mm inside diameter (Fig. 47.5). Concurrent monitoring of the respiratory rate and depth, and blood oxygenation remains essential. Administration of a partial antagonist (nalbuphine or butorphanol) will temporarily increase the rate and depth of respiration, reducing tissue oxygen requirements by reducing metabolism (Buss et al. 2018) and should be administered before oxygen supplementation (Haw et al. 2014). Oxygen supplementation is a practical solution to enhance pulmonary gas exchange in immobilized rhinoceroses and if used wisely one bottle is sufficient for many animals, therefore we recommend immediate intranasal or tracheal insufflation of oxygen (avoid high flow rates that can induce hypercapnia) in all recumbent rhinoceroses. Within a few minutes vital statistics will provide information about respiratory function and in most situations if all physiologic variables are satisfactory the flow can be reduced or discontinued (Table 47.3). A small percentage of animals, however, will develop a physiologic crisis where continued oxygen supplementation is critical. Aluminum oxygen bottles are now available that are small and lightweight, making them convenient for helicopter use.

Capnography offers a useful adjunct to monitoring with pulse oximetry and many portable devices combine measurement of end-tidal carbon dioxide with oxygen saturation of hemoglobin. Capnography is common practice in human anesthesia as an aid to confirm placement of endotracheal tubes and for early warning of hypoventilation. In expired respiratory gases, capnography provides a direct measure of CO_2 eliminated from the lungs. It also gives an indirect measure of CO_2 production in the tissues and circulatory transport of CO_2 to the lungs, but perhaps more importantly, it provides a good indirect measure of ventilation. Capnography is a rapid noninvasive technique that offers reliable information about tissue CO_2 production, circulation, pulmonary perfusion, and alveolar ventilation and respiratory patterns. It can be readily measured in recumbent rhinoceroses by placing a cut-off endotracheal tube in the nostril to allow for passage of air over the detector or sampling tube. For rhinoceros capture, capnography is gaining acceptance by practitioners as a tool to enhance anesthetic safety by offering a quick method to detect ventilatory or circulatory failure (Morkel et al. 2010).

Table 47.1 Suggested doses for chemical restraint of adult wild rhinoceroses including supplemental agents used for respiratory support.

	Immobilization			Respiratory Support	
Rhinoceros Species	**Protocol**	**Reversal**	**Reference Comments**	**Protocol**	**Reference Comments**
White rhinoceros	2–3.5 mg etorphine + 40–90 mg butorphanol + 25–50 mg midazolam IM dart Detomidine, medetomidine, midazolam or azaperone can be added to this mix (a) for better muscle relaxation and often better blood oxygenation DMM and (b) to speed induction DMMA	Naltrexone at 40 mg per mg M99 IV (full reversal) OR 2–2.5 mg diprenorphine per 1 mg M99 IV (reverses M99, but not BT)	Bush et al. (2005, 2011) Reduces respiratory depression, hypoxia, tachycardia, muscle rigidity, and tremors, but with slower induction and an animal that may stay on its feet Avoid butorphanol in combination with etorphine in rough terrain where a quick induction is safer	Produces an immobile rhinoceros in ~10 minutes and crating *without* partial opioid reversal In case of inadvertent overdose or cardiopulmonary suppression give diprenorphine to reverse the M99 while preserving the sedative effects of the BT	Bush et al. (2005, 2011, unpubl. data) Reverse part or all of opioid effects based on desired outcome
	3–4.5 mg etorphine + 40–60 mg azaperone (replace azaperone with 10–20 mg detomidine if no transport) IM dart Consider 5–20 mg midazolam slowly IV for muscle relaxation	*For crate reversal*: 2.4 mg M50:50 per 1 mg M99 plus 1–2 mg naltrexone IV if pushing NTX will be a relatively lively wake up and one must be adequately prepared (i.e., animal close to crate, rope properly attached to head, well organized team, etc.) *For field/boma reversal*: naltrexone at 40 mg per mg M99 IV (full reversal)	Kock et al. (1995, 2006); Rogers (1993a) Still considered standard translocation protocol Morkel (unpubl. data); Kock et al. (1995)	All white rhinoceroses: 20 mg butorphanol per 1 mg M99 (20 × M99 considered minimum dose for white rhinoceros, reduce to 10× or 15× if light) OR 20–40 mg nalbuphine IV OR 20–30 mg nalorphine IV (no longer available in Africa) and/or 1 mg M50:50	Kock et al. (1995, 2006); Hofmeyr & Morkel (unpubl. data) Butorphanol is being used with greater frequency for its partial agonist properties in white rhinoceroses across Africa
Black rhinoceros	4 mg etorphine + 40–60 mg azaperone (replace azaperone with 100 mg xylazine or 10 mg detomidine) + 5000 IU hyaluronidase IM dart Azaperone can be increased to 200 mg for a quicker induction if no transport. Can also combine azaperone with alpha-2 agonists.	*For crate reversal*: 20 mg butorphanol per mg M99 OR 1–1.8 mg M50:50 IV *For field/boma reversal*: Naltrexone at 40 mg per mg M99 IV (full reversal)	Morkel (1989) Higher M99 doses for *Diceros bicornis bicornis* Kock (1992): Kock et al. (2006) Hyaluronidase is always recommended to speed induction, especially as black rhinoceroses (unlike white rhinoceroses) often run themselves into trouble	Give 5 mg butorphanol IV to increase respiration and heart rate and lighten the anesthesia. A 10-mg dose will considerably lighten anesthesia and the rhinoceros may stand. Animal less likely to stand if kept in lateral position and the rhinoceros can be easily pushed down, if necessary. *Note*: Do not use the white rhinoceros respiratory protocol in black rhinoceroses as it will cause arousal *Instead*: 5 mg butorphanol IV, titrate to effect Important to have animal lateral and pump legs every 20 minutes	Hofmeyr (unpubl. data); Morkel (unpubl. data); Kock et al. (2006)
	2–2.5 mg thiafentanil (thianil) + 2–2.5 mg etorphine IM dart Can also use thiafentanil alone at etorphine doses (i.e. up to 5 mg) but watch the respirations	Same	Rogers (1993b)		

(*Continued*)

Table 47.1 (Continued)

	Immobilization			Respiratory Support	
Rhinoceros Species	**Protocol**	**Reversal**	**Reference Comments**	**Protocol**	**Reference Comments**
Greater one-horned rhinoceros	2–2.5 mg etorphine + 10 mg acepromazine IM dart OR 0.7 mg carfentanil	Diprenorphine at 2.5 mg per 1 mg M99 IV Naltrexone at 100 mg per 1 mg carfentanil IV	Dinerstein et al. (1990) One sudden arousal noted Induction times longer for breeding males	Cardiopulmonary depression not reported, 6–10 breaths per minute Surround target rhinoceros with 10–15 trained elephants	Dinerstein et al. (1990)
Sumatran rhinoceros Transport	2 mg etorphine + 80 mg azaperone + 5000 IU hyaluronidase IM dart OR Use M99:BT:MDZ	Naltrexone at 50 mg per mg M99 IM or IV	Author suggestion (extrapolated from captive animals)	Treat like black rhinoceroses; muscle rigidity and tremors common Use 5 mg midazolam to relax Use 5 mg butorphanol for partial reversal of respiratory depression	Radcliffe et al. (2002, unpubl. data)
	80 mg butorphanol + 80 mg azaperone IM dart OR Use MED:BT:MDZ (see Citino (2019) in Table 47.1)	Naltrexone at 2.5 mg per mg BT IM or IV	Use for compromised animal in snare	If rhinoceros is approachable give 25–40 mg butorphanol IV rather than via dart Sumatran rhinoceroses are easily tamed and can even be fed into a crate; a temporary boma can be erected to facilitate capture and crating	Radcliffe et al. (2002, unpubl. data); Citino (2019, unpubl. data)
	Long-acting tranquilizer for Crate transport and boma confinement: 50 mg zuclopenthixol acetate	No antagonist (see respiratory support)	40 mg azaperone Calm agitated rhinoceroses during crate transport 50 mg zuclopenthixol Repeat dose at boma offloading	10–20 mg biperidin OR 5 mg diazepam For control of undesirable extra-pyramidal effects	Candra (2018) Zuclopenthixol is the only LA tranquilizer that has been used in a Sumatran rhinoceros

BT, butorphanol; IM, intramuscular; IV, intravenous; M99, etorphine; MED, medetomidine; MDZ, midazolam; NTX, naltrexone.

Sources: Candra, 2018 Indonesian Rhino Capture Team Report; Morkel unpubl. data; Radcliffe unpubl. data; Bush M, Citino SB, Grobler D. 2005. Improving cardio-pulmonary function for a safer anesthesia of white rhinoceros (*Ceratotherium simum*): use of opioid cocktails to influence receptor effects. Proceedings of the American Association of Zoo Veterinarians, American Association of Wildlife Veterinarians, and American Zoo and Aquarium Association Nutrition Advisory Group, pp. 259–260; Bush M, Citino SB, Lance WR. 2011. The use of butorphanol in anesthesia protocols for zoo and wild mammals. In: *Fowler's Zoo and Wild Animal Medicine Current Therapy 7* (RE Miller, ME Fowler, eds.), pp. 596–603; Kock MD, Meltzer D, Burroughs R, eds. 2006. *Chemical and Physical Restraint of Wild Animals: A Training and Field Manual for African Species*. Zimbabwe Veterinary Association Wildlife Group and International Wildlife Veterinary Services; Rogers PS. 1993a. Chemical capture of the white rhinoceros (*Ceratotherium simum*) or 1993b. Chemical capture of the black rhinoceros (*Diceros bicornis*). In: *The Capture and Care Manual* (AA McKenzie, ed.). Pretoria: Wildlife Decision Support Service and South African Veterinary Foundation.

Table 47.2 Suggested opioid reversal protocols for walking, crate loading and transport of adult African rhinoceroses.

Method	Use	Reversal Drug or Opioid	Technique for Crating or Translocation
Butorphanol or nalorphine walking and crating method	For opiate (M99)-immobilized rhinoceroses	*White rhinoceroses* *Walking*: 10–20 mg BT per mg M99 Add 1–2 mg M50:50 for adult bulls for walking or crating (Hofmeyr) OR 1 mg M50:50 plus 20 mg nalorphine. Give further incremental 10–20 mg nalorphine IV up to 75 mg (Kock 2006) Nalorphine no longer available in Africa *Crating*: 10–20 mg BT per mg M99 OR Diprenorphine (M50:50) at 2.4× etorphine dose IV Add 1–2 mg naltrexone to prevent pushing in crate. Can combine the naltrexone with diprenorphine, expect a relatively lively wake up so be properly prepared with head rope, position of crate etc. *Black rhinoceroses* *Walking*: Start with 5 mg nalorphine IV Give incremental 5-mg doses every 5–10 minutes up to 20–40 mg (Kock et al. 2006) *Crating*: 15 mg butorphanol IV per 1 mg M99 with rhinoceros's head in crate door (Hofmeyr) Add 1–2 mg M50:50 to preclude pushing OR 10–20 mg nalorphine per 1 mg etorphine 1–2 mg M50:50 IV may be necessary to prevent pushing (Morkel) OR Blindfold with tight muslin cloth and wake up with 1–2 mg IV M50:50 Avoid myopathy in crating process by ensuring rhino does not squat in crate; use prodder plus repeat 1–2 mg M50:50 or 5 mg butorphanol doses	• Blindfold rhino. After cleaning eyes of mud or other debris, wrap a 4-m piece of muslin (mutton) cloth to cover eyes completely. Attach three zip ties to secure muslin cloth forward of posterior horn (Note: The blindfold option is really for black rhinoceroses and although it can be used on white rhinoceroses as well there is much less need for it). • Position the rhinoceros's head close to or inside crate door: black rhinoceroses very important to have head in door, white rhinoceroses not critical. Keep ears plugged until crated or leave in for transport. • Place head rope. Use 20 m of soft nylon behind posterior horn with knot on side of head, passing rope end through hole in crate. • Place break rope on rear leg just below hock. Use 8 m of nylon rope. • Position eight people on head rope and four people on break rope. Can use a 4 × 4 pick-up truck for back-up on rope if not enough people for head rope. • Reverse. Wait 90–120 seconds. Use electric prodder or water in ear to stimulate rhinoceros to stand if does not do so by itself. • Walk rhinoceros into crate by pulling on head rope. Slow rhinoceros with break rope or go slowly with head rope. Guide rhinoceros by ground personnel. Slide and secure pipes in crate (most crates have horizontal pipes, some only have the doors). • If black rhinoceros pushes in crate give 1 mg naloxone or 0.6–1.2 mg diprenorphine IV. If white rhinoceros pushes give 1–2 mg naltrexone IV. Use prodder on head or shoulders and not on hindquarters since rhinoceros will tend to push/squat more with stimulation of hindquarters.
Butorphanol alone crating method	For awake rhinoceroses in zoo or boma environments	*White rhinoceroses* 50 mg butorphanol IV (Radcliffe) *Black rhinoceroses* 25–50 mg butorphanol IV at time of crating for conditioned animals (Radcliffe) *Boma black rhinos*: Start with 10 mg butorphanol IV and increase in 5-mg increments (Hofmeyr)	Butorphanol is a useful agent for crating and transport of crate-conditioned rhinoceroses in zoological settings. Combine with azaperone as needed Butorphanol provides excellent sedation without concerns of excessive head pressing in crate and occlusion of nostrils in corner No reversal required for butorphanol once rhinoceros is in crate

(*Continued*)

Table 47.2 (Continued)

Method	Use	Reversal Drug or Opioid	Technique for Crating or Translocation
Diazepam: nalorphine crating method	For field-immobilized rhinoceroses	*White and black rhinoceroses* 10–15 mg diazepam IV 10 minutes before "waking" rhino with reversal protocol (Morkel) Use standard crating methodologies above for white and black rhinoceroses after giving diazepam	Give diazepam to recumbent rhinoceros and wait 10 minutes Use the same crating procedure as above using diprenorphine alone (white rhinoceroses) or nalorphine combined with diprenorphine (black rhinoceroses) This protocol eliminates much of the pushing often observed in the crate following diprenorphine or nalorphine reversal procedures Diazepam provides good sedation for ~8 hours, especially in white rhinoceroses
Etorphine: azaperone boma crating method	For loading and transport of boma rhinoceroses	*White rhinoceroses* *Boma, crating*: 1–2.5 mg etorphine IM (higher dose for adult bull) plus butorphanol at 10× M99 dose plus 20 mg (subadult) or 40 mg (adult) azaperone *Note*: Butorphanol can also be given at time of recumbency rather than in the original dart, particularly if rhinoceros requires medical care or other procedures under recumbency (Buss) *White or black rhinoceroses* 0.7–1.2 mg etorphine IM (Kock et al. 2006) OR 0.3 mg and 0.5 mg M99 for black and white rhinoceroses, respectively, without need to reverse *Crate sedation*: 0.05–0.15 mg etorphine IM plus 100–200 mg azaperone (can put in same syringe) IM or 10–30 mg diazepam IM (not in same syringe) *Transport*: 50–150 mg zuclopenthixol acetate IM	Use low doses of etorphine to crate rhinoceroses from boma; combine with azaperone in black rhinoceros; wave white flag on pole to lure rhinoceros into crate Etorphine is the only agent to calm an excitable rhinoceros inside a crate Butorphanol is replacing nalorphine and diprenorphine for both respiratory support and arousal for crate loading in both white and black rhinoceroses. Use small incremental doses (5 mg or less) in black rhinoceroses as reversal is more dramatic and produces a lively rhinoceros *For crate sedation*: nalorphine sedation wears off ~5 hours post-crating; thereafter give etorphine every 2 hours for duration of trip If rhinoceros is not excitable, give azaperone up to 200 mg per 6 hours *Note*: More etorphine is not effective within 3–4 hours of diprenorphine use and 8–24 hours following reversal with naltrexone Avoid perphenazine in white rhinoceros (if going to boma) as it causes anorexia; low dose OK if going straight to field

BT, butorphanol; IM, intramuscular; IV, intravenous; M50:50, ; M99, etorphine.

Sources: Buss, unpubl. data; Hofmeyr, unpubl. data; Morkel, unpubl. data; Radcliffe, unpubl. data; Kock MD, Meltzer D, Burroughs R, eds. 2006. *Chemical and Physical Restraint of Wild Animals: A Training and Field Manual for African Species*. Zimbabwe Veterinary Association Wildlife Group and International Wildlife Veterinary Services; Rogers PS. 1993a. Chemical capture of the white rhinoceros (*Ceratotherium simum*) or 1993b. Chemical capture of the black rhinoceros (*Diceros bicornis*). In: *The Capture and Care Manual* (AA McKenzie, ed.). Pretoria: Wildlife Decision Support Service and South African Veterinary Foundation.

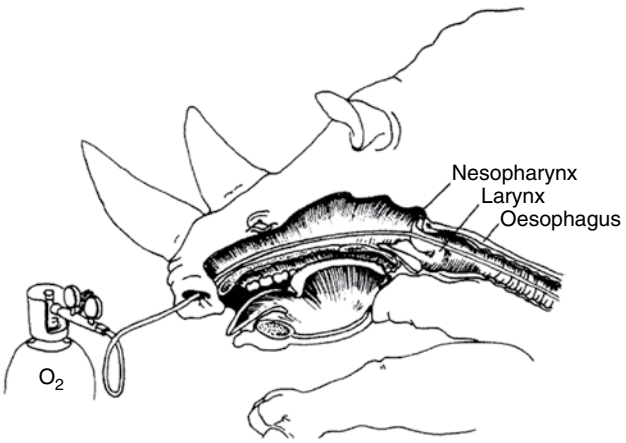

Figure 47.5 Illustration of the nasogastric tube tracheal insufflation technique for oxygen delivery in a recumbent white rhinoceros (*Ceratotherium simum*) under field conditions. Adapted from Bush et al. (2004), illustration courtesy of the South African Veterinary Association.

Table 47.3 Cardiopulmonary parameters in resting captive healthy white rhinoceroses (Citino & Bush 2007).

Parameter	Mean ± SD	Range
Heart rate (beats per minute)	39 ± 0.8	32–42
Respiration rate (breaths per minute)	19 ± 0.6	16–23
Rectal temperature (°C)	36.8 ± 0.1	36.6–37.2
Corrected indirect systolic pressure (mmHg)	160 ± 2.9	146–183
Corrected indirect diastolic pressure (mmHg)	104 ± 0.7	88–117
Corrected indirect mean pressure (mmHg)	124 ± 2.2	108–135
S_aO_2 (%)	97.2 ± 0.1	96.6–98
Arterial pH	7.391 ± 0.007	7.346–7.431
End-tidal CO_2 (mmHg)	45.1 ± 0.7	41.7–48
PaO_2 (mmHg)	98.2 ± 1.4	90.2–108.6
$PaCO_2$ (mmHg)	49 ± 0.9	44.4–53.7
Base excess (mmol/L)	3.5 ± 0.4	1.9–5.9
HCO_3^- (mmol/L)	29.3 ± 0.4	27.3–32.2

Body Temperature

Body temperature is an important variable and the best indicator of the degree of exertion and stress endured by the rhinoceros before induction. For every 1°C increase in body temperature above normal, there is an approximately 10% increase in oxygen consumption. A rhinoceros' body temperature varies slightly in relation to an inherent circadian rhythm, with a low at dawn and peak at dusk. Black rhinoceroses immobilized without excessive exertion have a rectal temperature of between 36.5 and 38.5°C, and anything over 38.5°C should be liberally doused with cool water. Young rhinoceroses tend to have a higher body temperature than adult rhinoceroses after running a comparable distance. Although drenching with water is important, it will not have a dramatic effect in lowering the core body temperature, as there is considerable thermal inertia in such a large mammal. It helps to fan the rhinoceros with branches or a portable leaf blower after the animal has been wetted with water. Holding leafy branches over the rhinoceros to provide shade can help lower the temperature, but it is important that people do not crowd around an immobilized rhinoceros and prevent air movement. A beach umbrella is the most efficient way to make shade: it is cheap, light, and easy to transport and can be folded to fit in the helicopter. A rhinoceros with a body temperature over 39°C must be processed quickly while a temperature greater than 41°C mandates immediate delivery of the antidote after dousing with water.

The rhinoceros, like other large mammals, is prone to hyperthermia during capture and translocation. The black rhinoceros appears to suffer a greater level of hyperthermia-related morbidity and mortality in the peri-capture period than the white rhinoceros. The goal of the capture team should be to minimize stress and exertion, and speed induction. The rise in body temperature can be documented while the animal is in recumbency following excessive exertion, however, a second phase apparently unrelated to the level of exertion appears to occur on crating. The mechanism of this hyperthermic response observed at variable periods after the rhinoceros enters the crate is not well understood, but could result from post-reversal agitation and muscle activity, a physiologic stress response, inadequate airflow inside the crate, or a combination of factors or mechanisms (Meyer et al. 2008). Simultaneous comparison of rectal and muscle temperatures in recumbent rhinoceroses demonstrates a 0.5°C (1°F) higher temperature in the muscle (Morkel et al. 2011). Since equilibration between rectal and muscle temperatures occurs slowly with time, a deep muscle thermistor can be a useful aid to measuring core body temperature. The probe is readily inserted into the dart site, but caution must be taken to avoid exposure to dangerous immobilizing drugs.

Additional options for temperature measurement in the rhinoceros include use of a handheld infrared thermometer (Fluke Hart Scientific Inc.) in the ear canal. In a recent pilot study deep-infrared ear temperatures were comparable to deep muscle temperatures, giving a reliable and rapid assessment of core body temperature. Surprisingly, the use of cotton or wool earplugs resulted

in ear canal temperature above deep muscle temperature and suggests that the inner ear in the rhinoceros may be an important site for cooling. In animals predisposed to hyperthermia (prolonged capture, high environmental temperature, etc.), rhinoceroses may benefit from removal of the earplugs and perhaps even application of some cooling mechanism to this site such as a cold pack (Morkel et al. 2010, 2011). Another tool that has been adopted to enhance temperature monitoring in rhinoceroses is the temperature microchip (LifeChip Inc.). After reaching equilibrium when implanted under the skin or into a muscle, chip temperatures were slightly lower than rectal temperatures.

Pulse and Blood Pressure

Heart rate is best obtained using a stethoscope while the pulse is readily palpable on the inside of the ear (medial auricular artery) or under the base of the tail (caudal artery). Pulse quality should be evaluated subjectively and compared with the pulse oximeter reading. It is often quite easy to visualize heart compressions by watching the chest wall or by feel with a hand placed over the cardiac window. The heart rate is usually 55–80 beats per minute (bpm) although it will be higher in rhinoceroses that have undergone marked exertion, especially in young animals (up to 140 bpm). Cardiovascular function and peripheral perfusion are assessed by capillary refill time and is measured by blanching the rhinoceros' gum for several seconds and then releasing. The observed delay or refill time should not exceed 2 seconds.

Hypertension is prevalent under etorphine anesthesia in black and white rhinoceroses (LeBlanc et al. 1987; Heard et al. 1992; Hattingh et al. 1994). Reports in white rhinoceroses anesthetized under field conditions noted reduction in blood pressures when azaperone tartrate (Stresnil) replaced fentanyl in etorphine-based combinations, an effect observed despite the higher dose of etorphine used in the cocktails containing azaperone (Hattingh et al. 1994; Buss et al. 2016). Sympathetic activation during immobilization of rhinoceroses is a very important factor that is not fully appreciated and may be the greatest cause of hypoxemia during etorphine immobilization (Meyer, pers. comm., 2020). Increased sympathetic nervous system action, peripheral vasoconstriction, and hypoxemia are purported factors in etorphine-induced hypertension in both rhinoceroses and equids (Daniel & Ling 1972; Heard et al. 1992). Opioid-related hypoxemia may induce sympathetic system stimulation and hypertension. Elevated heart rate is a good indicator of hypoxia; once hypoxia resolves the sympathetic response and associated hypertension disappear.

Rhinoceros Anesthesia in Captivity

Guidelines for Anesthesia of Captive Rhinoceroses

The large size of the rhinoceros belies an unexpected sensitivity to the opioid class of pharmacologic agents (Raath 1999). Surprisingly, the same dose of carfentanil citrate used to immobilize a 20-kg blackbuck (*Antelope cervicapra*) would also fully immobilize a 2200-kg white rhinoceros (*Ceratotherium simum*), making the rhinoceros over 100 times more opioid sensitive per unit mass than the average artiodactylid. This inordinate sensitivity of the Rhinoceros family to the opioid class – while responsible for the undesirable changes observed in cardiopulmonary function – also makes it possible to adapt less potent mixed agonist-antagonist opioid agents into anesthetic protocols for both captive and wild rhinoceroses (Radcliffe et al. 2000a; Walzer et al. 2000; Bush et al. 2005).

Planning for anesthetic events should include preparation of the subject and environment where these variables can be controlled. Depending on the purpose for anesthesia it is generally desirable to fast the animal for 12–24 hours prior to anesthesia (Radcliffe et al. 2000b). However, fasting is certainly not essential, as evidenced by the many successful field operations where capture of wild rhinoceroses is conducted in the absence of preanesthetic fasting. Water access should be denied for at least 12 hours and all water sources removed from the environment prior to drug delivery to prevent animals from losing consciousness while in a mud wallow or water pond. Both passive and active regurgitation of stomach contents are known, with the latter being very rare but quite spectacular (Radcliffe et al. 1998; Raath 1999). Passive regurgitation is common in immobilized rhinoceroses presumably secondary to drug- or hypoxemia-induced relaxation of the cardiac sphincter (Fig. 47.6). Because of the risks of regurgitation and inhalation pneumonia, great care must be taken with positioning of the head and nostrils, especially with animals in lateral recumbency.

Habitual patterns of behavior are important aspects of captive rhinoceros husbandry, facilitating close medical management. Anesthesia techniques should be adapted as part of these conditioning protocols. Regular visits by animal health staff to rhinoceros barns or bomas for acclimatization to the sights, sounds, and smells of the veterinary profession will help limit the stress of such procedures. In boma situations it is helpful to learn the nature of each animal, including its likes and dislikes, while also listening carefully to the keeper in charge of caring for the animal.

Figure 47.6 Passive regurgitation from the nares of a black rhinoceros (*Diceros bicornis*) forcefully expelled at expiration. The rhinoceros is positioned in sternal recumbency to help the reflux drain away from the airway.

Figure 47.7 Typical induction posture in adult white rhinoceros (*Ceratotherium simum*) under the effects of etorphine, illustrating characteristic head elevation, raised hackney action of forelimbs, and muscle rigidity. Image courtesy of Rolfe Radcliffe, Living Fossil Foundation.

African Rhinoceros Captive Anesthetic Regimens

White Rhinoceros (*Ceratotherium simum*)

The adult white rhinoceros is large and generally placid in captivity. Anesthesia with potent opioids is often associated with marked hypermetria, muscle rigidity, trembling, head shaking, and limb paddling (Fig. 47.7). These effects are undesirable and can be prevented by preanesthetic administration of the sedative or tranquilizer component of the cocktail and butorphanol post induction. In captive animals, initial dosing with IM azaperone or detomidine 20–30 minutes prior to induction with etorphine helps preclude muscle spasms and rigidity. With wild rhinoceroses, positioning in sternal recumbency until complete relaxation is achieved was deemed important in field practice (Kock et al. 1995).

Mixtures of etorphine or carfentanil combined with a sedative are standard agents for anesthesia of the captive white rhinoceros (Table 47.4). Doses ranging from 0.8 mg up to 3 mg of etorphine and 1.2 mg carfentanil are common (Heard et al. 1992; Walzer et al. 2000), however, supplemental opioids traditionally administered to extend anesthesia should be avoided as they have been linked to profound increases in pulmonary artery pressure and cardiac output, and decrease in arterial oxygen (Boesch et al. 2018). Following immobilizing doses of etorphine or carfentanil, other agents provide additional muscle relaxation and a deeper plane of anesthesia, including IV alpha-2 agents, propofol, guaifenesin, ketamine, and midazolam (Klein et al. 1997; Zuba & Burns 1998; Walzer et al. 2000; Kock et al. 2006). Muscle relaxation is critical for deep ventilation and to preclude the oxygen depletion and hyperthermia from muscle tremors inherent with the use of potent opioids. However, if used alone, these drugs may mask sympathetic activation that predisposes immobilized rhinos to tachycardia, cardiac hypoxia, and increased cardiac workload. Butorphanol does a good job of countering the adverse effects of sympathetic activation in white rhinoceroses.

Lower opioid doses are indicated in zoo-conditioned animals, yet the potent opioids are still associated with significant cardiopulmonary changes, especially as procedure length increases (Heard et al. 1992). One captive white rhinoceros immobilized with etorphine remained hypoxemic despite maintenance of inhalation anesthesia using intermittent partial positive pressure ventilation (Cornick-Seahorn et al. 1995). Hypertension is common while hypoventilation, pulmonary hypertension and shunting, and atelectasis induce hypoxia and hypercapnia (Heard et al. 1992; Bush et al. 2004).

Butorphanol combinations are replacing the use of more potent opioids for rhinoceros anesthesia in many zoological settings as safe and reliable anesthetic planes can be achieved for most procedures, including surgery (Radcliffe et al. 2000a–c). While not appropriate for all applications (i.e., fractious, nonconditioned animals or those with access to large areas) butorphanol combinations are highly effective. The author has used a mixture of butorphanol and azaperone for standing sedation and recumbent anesthesia in all four rhinoceros species maintained in captivity (white, black, greater one-horned, and Sumatran) with safe, predictable results (Radcliffe et al. 2000a,c; Radcliffe & Morkel 2007). Butorphanol doses for white rhinoceros range from 50 to 120 mg for an adult and from 10 to 20 mg for a calf or juvenile animal while azaperone doses range from 100 to 160 mg for an adult with supplemental doses given up to a maximum of 300 mg (Tables 47.4 and 47.5). IV butorphanol supplementation is effective at inducing

Table 47.4 Suggested doses for chemical restraint of adult captive rhinoceroses producing anesthetic planes from sedation to recumbency.

	Standing Sedation			Recumbency		
Rhinoceros Species	**Protocol**	**Reversal**	**Reference Comments**	**Protocol**	**Reversal**	**Reference Comments**
White rhinoceros	50–70 mg butorphanol + 100 mg azaperone IM hand-injection plus CRI	Naltrexone at 2.5 mg per mg BT IM or IV	Radcliffe et al. (2000a,b) Use CRI in long procedures	70–120 mg butorphanol + 100–160 mg azaperone IM hand-injection	Naltrexone at 2.5 mg per mg BT IM or IV	Radcliffe et al. (2000a) Supplemental IV dosing or CRI
	120–150 mg butorphanol + 5–7 mg medetomidine IM dart (Give 1–2 mg nalorphine IV to keep standing)	Naltrexone at 1 mg per mg BT Atipamezole at 5 mg per mg MED	Citino (2008)	120–150 mg butorphanol + 5–7 mg medetomidine (IM dart; recumbency ~20 minutes)	Naltrexone at 1 mg per mg BT Atipamezole at 5 mg per mg MED	Citino (unpubl. data) Improved analgesia for surgery
	0.8–1.5 mg etorphine IM dart	Naltrexone at 40 mg per mg M99	Portas (2004)	2–3 mg etorphine + 20–40 mg azaperone IM dart	Naltrexone at 40 mg per mg M99	Portas (2004)
				1.2 mg carfentanil IM dart	Naltrexone at 100 mg per mg M99	Portas (2004)
Black rhinoceros	25–50 mg butorphanol IV or IM hand-injection	Naltrexone at 2.5 mg per mg BT IM or IV	Radcliffe et al. (2000c; unpubl. data) Use for subadults and crating	1–1.5 mg etorphine + 100 mg azaperone IM hand-injection	Naltrexone at 50 mg per mg M99 half IV, half IM	Radcliffe (unpubl. data) Lower M99 doses with hand-injection
	1.5–2 mg etorphine + 2–3 mg medetomidine + 15–20 mg midazolam (Give 1–2 mg nalorphine IV to keep standing) IM dart	Naltrexone at 30 mg per mg M99 Atipamezole at 5 mg per mg MED	Citino (2019)	1.5–2 mg etorphine + 2–3 mg medetomidine + 15–20 mg midazolam (IM dart; recumbency ~15 minutes)	Naltrexone at 30 mg per mg M99 Atipamezole at 5 mg per mg MED	Citino (unpubl. data) Enhanced analgesia for dental surgery; midazolam to reduce aggression on recovery
	2–2.5 mg etorphine + 10 mg detomidine + 15 mg butorphanol IM dart	Naltrexone at 40 mg per mg M99 Atipamezole at 5 mg per mg DET	Portas (2004)	2.5–3 mg etorphine + 60 mg azaperone IM dart	Naltrexone at 20–40 mg per mg M99	Portas (2004)

Greater one-horned rhinoceros	100 mg butorphanol + 100 mg azaperone IM hand-injection	Naltrexone at 2.5 mg per mg BT IM or IV	Radcliffe & Lung (unpubl. data)	3.5–3.8 mg etorphine + 14 mg detomidine + 400 mg ketamine IM pole-syringe	150–300 mg naltrexone half IV, half IM No reversal DET	Atkinson et al. (2002)
	Medetomidine (4 μg/kg) Butorphanol (45–60 μg/kg) Midazolam (15–22 μg/kg)	Naltrexone at 1 mg per mg BT Atipamezole at 5 mg per mg MED	Citino (2019)	Etorphine (2 μg/kg) Medetomidine (3–4 μg/kg) Ketamine (0.1–0.15 mg/kg)	Naltrexone at 30 mg per mg M99 Atipamezole at 5 mg per mg MED	Citino (2019)
Sumatran rhinoceros	25–40 mg butorphanol IM hand-injection *Note*: Sumatran rhinoceroses are easily conditioned to chute restraint and can be hand-injected. Even wild rhinoceroses can be tamed quickly with feed and walked into a crate without use of drugs!	Naltrexone at 2.5 mg per mg BT IM or IV	Radcliffe et al. (2002) Use azaperone in longer procedures	30–50 mg butorphanol + 50–60 mg azaperone IM hand-injection	Naltrexone at 2.5 mg per mg BT IM or IV	Radcliffe et al. (2002) Higher doses for recumbency
				1 mg etorphine + 60 mg azaperone IM hand-injection	Naltrexone at 50 mg per mg M99 half IV, half IM	Radcliffe (unpubl. data) Azaperone 20 minutes prior to M99 or supplement midazolam
				10 mg butorphanol + 10 mg detomidine IM dart Wait 20 minutes 1.2 mg etorphine 5 mg acepromazine IM dart	150 mg naltrexone IV + 20 mg atipamezole IV	Walzer et al. (2010) 530-kg adult male; ketamine boluses (50 mg each) to extend anesthesia
	Adult female (wt. = 625 kg): 2.5 mg medetomidine (4 μg/kg) 20 mg butorphanol (31 μg/kg)	Naltrexone at 1 mg per mg BT Atipamezole at 5 mg per mg MED	Citino (2019) Excellent standing sedation for uterine endoscopy	Adult male: 4–5 mg medetomidine 40–50 mg butorphanol 20 mg midazolam	Naltrexone at 1 mg per mg BT Atipamezole at 5 mg per mg MED	Citino (2019) Combination for electroejaculation; supplement with glyceryl guaiacolate, medetomidine and ketamine

BT, butorphanol; CRI, constant rate infusion; DET, detomidine; IM, intramuscular; IV, intravenous; M99, etorphine; MED, medetomidine.

Sources: S.B. Citino, pers. comm., 2008, 2019. Use of medetomidine in chemical restraint protocols for captive African rhinoceroses. Joint Proceedings of the American Association of Zoo Veterinarians and the Association of Reptilian and Amphibian Veterinarians, Los Angeles, CA, pp. 108–109; Radcliffe, unpubl. data; Atkinson MW, Bruce H, Gandolf AR, Blumer ES. 2002. Repeated chemical immobilization of a captive greater one-horned rhinoceros (*Rhinoceros unicornis*), using combinations of etorphine, detomidine, and ketamine. Journal of Zoo and Wildlife Medicine 33(2):157–162; Portas TJ. 2004. A review of drugs and techniques used for sedation and anaesthesia in captive rhinoceros species. *Australian Veterinary Journal* 82(9):542–549; Walzer C, Goritz F, Hermes R, Nathan S, Kretzschmar P, Hildebrandt T. 2010. Immobilization and intravenous anesthesia in a Sumatran rhinoceros (*Dicerorhinus sumatrensis*). Journal of Zoo and Wildlife Medicine 41:115–120.

Table 47.5 Suggested doses for immobilization and anesthesia of rhinoceros calves in both captive and wild environments.

	Captive Calves			Wild Calves		
Rhinoceros Species	**Protocol**	**Reversal**	**Reference Comments**	**Protocol**	**Reversal**	**Reference Comments**
White rhinoceros	10–20 mg butorphanol IV for 66–159-kg calf (dose 0.13–0.15 mg/kg IV)	Naltrexone at 5 mg per mg BT	Gandolf et al. (2006) Heavy sedation Light anesthesia Mild resedation noted 8 hours post-reversal in one calf	*Calf*: 0.1–1 mg etorphine *Juvenile*: 1–2.5 mg etorphine *Subadult*: 2.5–3.5 mg etorphine (Note: above ranges represent calves of all sizes) *Note*: All white rhinoceroses, including calves, get IV BT at 10–20× the 1-mg M99 dose as soon as possible on recumbency to provide respiratory support OR 1 mg M50:50 plus 10 mg nalorphine IV	Diprenorphine at 2.5 mg IV per 1 mg M99 for transport Naltrexone at 40 mg per 1 mg M99	Kock et al. (2006) from South African National Parks Note: Always dart mother rhinoceros 30–60 seconds before calf (primarily for black rhinoceroses who easily split; in the case of white rhinoceroses, the calf rarely leaves the mother's side so one can wait longer or until mother goes down before darting the calf)
	2.5–5 mg butorphanol + 1.5–1.8 mg detomidine IM for 69–122-kg calf (dose 0.03 mg/kg BT plus 0.07 mg/kg DET)	Naltrexone at 4 mg per 1 mg BT Yohimbine at 0.125 mg/kg	Gandolf et al. (2006) Surgical anesthesia	*Calf*: 0.1–1 mg etorphine + 5–20 mg azaperone Subadult: 2.5–3.5 mg etorphine + 30–60 mg azaperone	Diprenorphine at 3 mg IV per 1 mg M99	Rogers (1993a)
Black rhinoceros	25 mg butorphanol IV for ~500-kg subadult calf	Naltrexone at 5 mg per 1 mg BT	Radcliffe et al (2000c) Heavy standing sedation	*Calf*: 0.1–1 mg etorphine Subadult: 2.5–3.5 mg etorphine	Naltrexone at 40 mg IV per 1 mg M99	Kock et al. (2006) from SANP
				Calf: 0.1–1 mg etorphine + 10–50 mg azaperone Subadult: 1.75–3.5 mg etorphine + 100 mg azaperone	Diprenorphine at 3 mg IV per 1 mg M99	Rogers (1993b); Kock et al. (2006) Note: Always dart mother rhinoceros 30–60 seconds before calf
				Note: Do not use the M50:50 plus nalorphine protocol in black rhinoceroses as it will cause arousal Instead: 5 mg butorpahnol or nalorphine IV; titrate to effect		
Greater one-horned rhinoceros	Butorphanol IV or IM Use white rhino as model	Naltrexone at 5 mg per 1 mg BT	Author suggestion based on use in African rhinoceros calves	*Calf*: 0.5–1 mg etorphine + 5 mg acepromazine *Subadult*: 2–2.5 mg etorphine + 10 mg acepromazine	Diprenorphine at 2.5 mg IV per 1 mg M99	Dinerstein et al. (1990) Same dose used for adult/subadult

BT, butorphanol; DET, detomidine; IM, intramuscular; IV, intravenous; M50:50, Diprenorphine; M99, etorphine.

Sources: Dinerstein E, Shrestha S, Mishra H. 1990. Capture, chemical immobilization, and radio-collar life for greater one-horned rhinoceros. *Wildlife Society Bulletin* 18(1):36–41; Atkinson MW, Bruce H, Gandolf AR, Blumer ES. 2002. Repeated chemical immobilization of a captive greater one-horned rhinoceros (*Rhinoceros unicornis*), using combinations of etorphine, detomidine, and ketamine. *Journal of Zoo and Wildlife Medicine* 33(2):157–162; Gandolf AR, Wolf TM, Radcliffe RW. 2006. Serial chemical restraint for treatment of decubitus ulcers in two neonatal white rhinoceroses (*Ceratotherium simum*). *Journal of Zoo and Wildlife Medicine* 37(3):387–392; Kock MD, Meltzer D, Burroughs R, eds. 2006. *Chemical and Physical Restraint of Wild Animals: A Training and Field Manual for African Species*. Zimbabwe Veterinary Association Wildlife Group and International Wildlife Veterinary Services.

recumbency in white rhinoceroses after initial drug delivery, if needed and desirable. IV dosing of azaperone without prior sedation has been associated with adverse extrapyramidal reactions in the horse and white rhinoceros, and should be avoided (Radcliffe et al. 2000a). Combinations of butorphanol and medetomidine offer another safe alternative that can produce dose-dependent anesthesia from standing sedation to surgical anesthesia for shorter procedures (Morkel & Nel 2019).

Inhalation anesthesia is possible in captive rhinoceroses where more invasive procedures requiring surgery or longer anesthesia times are indicated. Intubation in the rhinoceros is accomplished by hand or with an endoscope to guide placement of the endotracheal tube or a guide catheter into the airway. Unlike the horse, rhinoceroses have a unique, blind diverticulum in the dorsocaudal pharynx above the glottis. The anesthetist must be careful not to pass the tube into the diverticulum during intubation (Radcliffe et al. 1998).

A white rhinoceros was safely anesthetized to treat a surgical colic by sedation with butorphanol (80 mg) and detomidine (50 mg) followed by IV glyceryl guaiacolate (50 g) and three boluses of ketamine (200 mg per bolus) for induction (Valverde et al. 2010). Anesthesia was maintained for an additional 6 hours using isoflurane in oxygen delivered at 1–2% using a circle breathing system. Positioning for surgery and recovery were challenging, but made possible through the use of inflatable mats and the expertise of the local fire department. Although the rhinoceros eviscerated 3 days post surgery, the anesthesia and recovery were considered a success. Inhalation anesthesia together with a sling system facilitated laparoscopic-assisted transvaginal oocyte recovery in a black rhinoceros (Portas et al. 2006). Abdominal laparoscopy with insufflation of the abdomen is possible in rhinoceroses without general anesthesia by using a standing approach (Radcliffe et al. 2000b).

Black Rhinoceros (*Diceros bicornis*)

Black rhinoceroses appear predisposed to excitation during induction with etorphine, especially with remote drug delivery in zoological environments (Portas 2004). Using appropriate human safety practices, the stress of darting can be avoided by hand-injection, thereby alleviating much of the undesirable excitatory phase black rhinoceroses experience while also significantly reducing the total dose of opioid agents required (Radcliffe & Morkel 2007; Table 47.4). In bomas, to limit the "undesirable excitatory phase", great care should be taken to minimize the number of people and unusual objects close to the boma. Noise and movement should be avoided and, once recumbent, the rhinoceros' eyes should be covered and ears blocked as soon as possible. Significant induction risks include lacerations, limb and foot injuries, head trauma, damage to nasal sinuses, horn avulsion, and even death. With careful animal conditioning and procedure planning the risks of induction excitation are easily minimized. Likewise, antagonism of narcotic anesthesia in the black rhinoceros is characterized by rapid and powerful recoveries mandating extra care; never stand in front of a narcotized rhinoceros as arousal is often sudden and unpredictable (Kock et al. 2006).

As in the other rhinoceros species, potent opioids (primarily etorphine) have historically been used for anesthesia of captive black rhinoceroses with predictable results (Portas 2004). Zoo-conditioned animals require much lower doses of etorphine (1–1.5 mg) than their wild counterparts, especially when administered by hand-injection or pole-syringe (Table 47.4). Butorphanol alone or in combination with azaperone or detomidine HCl (Dormosedan) has also been used in the black rhinoceros, although its use is primarily limited to subadult animals, crating and translocation procedures, or well-conditioned animals since black rhinoceroses are easily excitable and may override drug effects (Radcliffe et al. 2000c). In addition to reversing the respiratory depressant effects at the μ receptors, butorphanol also antagonizes the powerful μ sedative effects, thereby greatly lightening anesthesia. For butorphanol use in the black rhinoceros, expect light planes of anesthesia and the need for frequent redosing. A more thorough discussion of mixed agonist-antagonist opioid cocktails and newer alpha-2 agents for use in both captive and field immobilization protocols for the African rhinoceroses can be found in the new techniques section of this chapter and in Tables 47.1 and 47.4.

Asian Rhinoceros Captive Anesthetic Regimens

Indian or Greater One-horned Rhinoceros (*Rhinoceros unicornis*)

Despite the common occurrence of Indian rhinoceroses in zoological parks and a propensity for foot problems necessitating chronic care, few published accounts of anesthesia in captive greater one-horned rhinoceroses exist (Atkinson et al. 2002; Portas 2004). One report combined injectable and inhalation anesthesia in a female *Rhinoceros unicornis* for ovariohisterectomy using etorphine and isoflurane in oxygen. The 7-hour long anesthesia (much of it in dorsal recumbency) was considered effective despite the animal succumbing to postsurgical complications (Klein et al. 1997). The most complete summary of captive anesthesia in this species, however, describes serial opioid-based anesthesia to facilitate long-term medical foot care in one animal. A combination of etorphine-detomidine (3–3.6 and 10–14 mg IM,

respectively) was given by projectile dart or etorphine-detomidine-ketamine (3.5–3.8, 14, and 400 mg IM, respectively) administered by pole-syringe (Atkinson et al. 2002). Use of the pole-syringe for drug delivery was preferred because darting was limited by a small target area among the peculiar anatomic neck folds and by drug selection for small dart volumes. While both drug combinations proved efficacious, subjective assessment suggested that the etorphine-detomidine-ketamine protocol produced more rapid induction, lowered the need for supplemental ketamine, and shortened reversal times (Atkinson et al. 2002).

The author has used butorphanol and azaperone (100 mg of each drug mixed in a syringe and given by hand-injection) to induce standing sedation in the Indian rhinoceros (Radcliffe & Lung, unpubl. data). A combination of butorphanol and detomidine (120 and 80 mg, respectively) produced sternal recumbency for surgical repair of a rectal prolapse (Bertelsen et al. 2004). As in the white rhinoceros, these protocols provide adequate muscle relaxation, sedation, and analgesia while being completely reversible with the pure opioid antagonists naltrexone or naloxone hyrdochloride (Narcan). Naltrexone is preferred unless short immobilization intervals are anticipated since renarcotization is common using naloxone alone; naloxone provides complete reversal for a short duration (~30–60 minutes) and is only suggested if repeat procedures are planned for the same day (Gandolf et al. 2000; Radcliffe et al. 2000a; Bertelsen et al. 2004; Portas 2004).

Javan or Lesser One-horned Rhinoceros (*Rhinoceros sondaicus*)

Rhinoceros sondaicus is the only rhinoceros not presently represented by captive specimens and was only extraordinarily displayed in zoological gardens during the seventeenth, eighteenth, and nineteenth centuries (Rookmaaker 1998). Although historical records indicate that at least 22 Javan rhinoceroses were captured between 1647 and 1939, only four survived long enough to reach zoo exhibits in Adelaide, Calcutta, and London (Rookmaaker 1998). The entire surviving wild population of Javan rhinoceroses can be found in Ujung Kulon National Park in West Java ($n \approx 72$). No accounts of Javan rhinoceros anesthesia exist, but techniques presumably would be analogous to the approaches used for the Sumatran rhinoceros (*Dicerorhinus sumatrensis*) or greater Asian one-horned rhinoceros (*Rhinoceros unicornis*), with size difference being a notable exception.

Sumatran Rhinoceros (*Dicerorhinus sumatrensis*)

Few reports of Sumatran rhinoceros anesthesia exist since captive specimens are rare. Etorphine (0.98–1.23 or 1 mg) combined with acepromazine (PromAce, 4–5 mg) or azaperone (60 mg) has been used to anesthetize captive Sumatran rhinoceroses (Portas 2004; Radcliffe & Morkel 2007). One adult male was immobilized on two occasions using a two-stage darting protocol. The first doses were considered inadequate and the authors subsequently recommended 10 mg of butorphanol plus 10 mg of detomidine IM followed 20 minutes later with 1.2 mg of etorphine and 5 mg of acepromazine IM, plus 50-mg supplemental doses of ketamine IV to extend the anesthesia period (Walzer et al. 2010). Darting of this animal likely contributed to long induction times (up to 40 minutes); Sumatran rhinoceroses are easily conditioned for hand-injection in a chute. As with the African species, muscle rigidity and cardiopulmonary depression are common with use of the potent opioids and preanesthetic administration of a tranquilizer is prudent to limit muscle tremors and improve respiratory function. Total azaperone doses should be kept to 100 mg or less as ataxia has been noted on recovery with higher doses in this species. Butorphanol has been combined with detomidine for standing sedation while the author routinely uses a mixture of butorphanol and azaperone for standing sedation and full recumbent procedures (Table 47.4; Radcliffe et al. 2002).

As with the African species, butorphanol combinations are preferred in captive Sumatran rhinoceroses to preclude the adverse cardiopulmonary changes associated with the use of more potent opioids. For adult animals butorphanol at a dose of 60–80 μg/kg with azaperone at 80–100 μg/kg and ranges of 30–50 mg and 50–60 mg butorphanol and azaperone, respectively, is recommended, with higher butorphanol doses being used on occasion to induce recumbency. Antagonism of the butorphanol effects is accomplished with naltrexone at a dose of 2.5 times the dose of butorphanol (Table 47.4; Radcliffe et al. 2002). Other tranquilizers may be used in place of azaperone such as the alpha-2 agonists, but care should be exercised as hypoxemia has been reported with use of these sedatives. Local anesthetics may facilitate invasive procedures, however, use of more potent narcotics such as etorphine or other pharmacologic agents such as ketamine and medetomidine may be indicated to induce surgical anesthesia.

New Captive Anesthesia Techniques

Although much has been learned about rhinoceros anesthesia, limitations still hinder safe and reliable procedures for these large mammals, especially where prolonged recumbency or surgery is required (Heard et al. 1992; Klein et al. 1997). Standing restraint where possible using mixed agonist-antagonists shows promise (Radcliffe et al. 2000a,b).

For the black rhinoceros, where potent opioids are still often preferred over mixed agonists, challenges include marked respiratory depression, inadequate muscle relaxation, need for frequent redosing, and incomplete analgesia in painful procedures. The incorporation of the potent alpha-2 agonist medetomidine with etorphine or butorphanol enhances sedation and analgesia in captive rhinoceroses (Citino 2008). Because alpha-2 agonists exacerbate respiratory depression and hypotension, contribute to dehydration, and alter thermoregulatory mechanisms, they must be used with caution in rhinoceroses of unknown health status, especially old and debilitated animals. However, under captive conditions where the health of an animal is known and a specific type of anesthesia is desirable, alpha-2 agents are effective supplements.

For the black rhinoceros, medetomidine (2–3 mg representing 2–2.9 μg/kg IM, 20 mg/mL solution) is combined with etorphine (1.5–2 mg representing 1.5–1.7 μg/kg IM; Citino 2008) and given by dart. The investigators were able to begin safe animal manipulations at approximately 9 minutes with full recumbency achieved in 15 minutes. This combination facilitated very painful procedures, including molar extraction and foot surgery, with the additional supplement of an IV guaifenesin-ketamine drip (1 g of ketamine in 1 L 5% guaifenesin solution) to enhance peripheral analgesia. Relaxation was excellent, with easy access to the oral cavity for dental surgery. Physiologic parameters were considered normal with concomitant nasal oxygen insufflation. Recovery from anesthesia was smooth and rapid, with no evidence of resedation or renarcotization using naltrexone at 30 mg per milligram of etorphine and atipamezole HCl (Antisedan) at 5 mg per milligram of medetomidine.

For white rhinoceros, where butorphanol has proven so effective in captive settings, the same investigator is using medetomidine (5–7 mg representing 2.47–2.81 μg/kg IM) and butorphanol (120–150 mg IM representing 62.5–64.9 μg/kg IM) to provide enhanced muscle relaxation and analgesia properties (Citino 2008). The animals can be manipulated within approximately 11 minutes of IM drug delivery with full recumbency in 20 minutes. The addition of medetomidine into these protocols has significantly improved muscle relaxation and analgesia properties for such painful ophthalmic procedures as eye enucleation and conjunctival flap surgery. As with the black rhinoceros, a 5% guaifenesin-ketamine drip was deemed useful for long procedures and to enhance peripheral analgesia. Antagonism was complete using naltrexone at 1 mg per milligram of butorphanol (204–262 μg/kg naltrexone) and atipamezole at 5 mg per milligram of medetomidine (25.4–31.2 μg/kg atipamezole).

Rhinoceros Anesthesia in the Wild

Guidelines for Anesthesia of Wild Rhinoceroses

Field anesthesia of Asian and African rhinoceroses is often undertaken to facilitate urgent conservation actions such as horn trimming, ear-notching, microchip application, radio-collaring, and horn transmitter implantation or translocation to protected areas (Dinerstein et al. 1990; Flamand et al. 1984). Ideally, rhinoceros capture operations should be conducted when environmental temperatures are lower than 25°C, usually in the early morning or late afternoon. Darting free-ranging rhinoceroses when ambient temperatures are high increases the risk of elevated body temperatures and associated physiological stress. If working in the late afternoon, a rhinoceros should not be darted unless there is sufficient daylight remaining (an hour is a minimum time to process the animal and deal with potential problems; Rogers 1993a,b). If a rhinoceros has run hard enough for its skin to become dark with sweat, the rhinoceros' body temperature will often exceed 39°C. Such an animal should not be darted or if it has already been darted, it must be drenched with water and processed quickly. If the temperature of an immobilized rhinoceros rises above 41°C, the antidote should be administered immediately and the animal released.

With good dart placement, recumbency should follow within 4–6 minutes post drug delivery (Morkel 1989, 1994; Kock et al. 2006). Induction is usually quicker in young rhinoceroses and longer in large bulls and heavily pregnant cows. If there are no signs at about 6 minutes, the rhinoceros should be darted again. Induction times of less than 3 minutes may indicate an overdose and it is important to get to such an animal quickly so that the respiration and other vital functions can be monitored; oxygen combined with a partial antagonist should be given, if necessary. In protocols incorporating thiafentanil (not recommended in white rhinoceroses), rapid inductions are expected and less of a concern. IV opioid use should be avoided due to risks of apnea, but if necessary give the opioid slowly while keeping a close eye on respiration. For the same reason caution must be exercised when giving midazolam or alpha-2 agonists by the IV route.

As a rhinoceros becomes affected by etorphine, its pace shortens, the forelegs are lifted higher in a classic "Hackney gait", and the head is elevated (Fig. 47.8). The rhinoceros then starts to blunder through bushes and slows down before going into lateral or sternal recumbency. In rough terrain rhinoceroses often run downhill once they are heavily narcotized and may easily injure

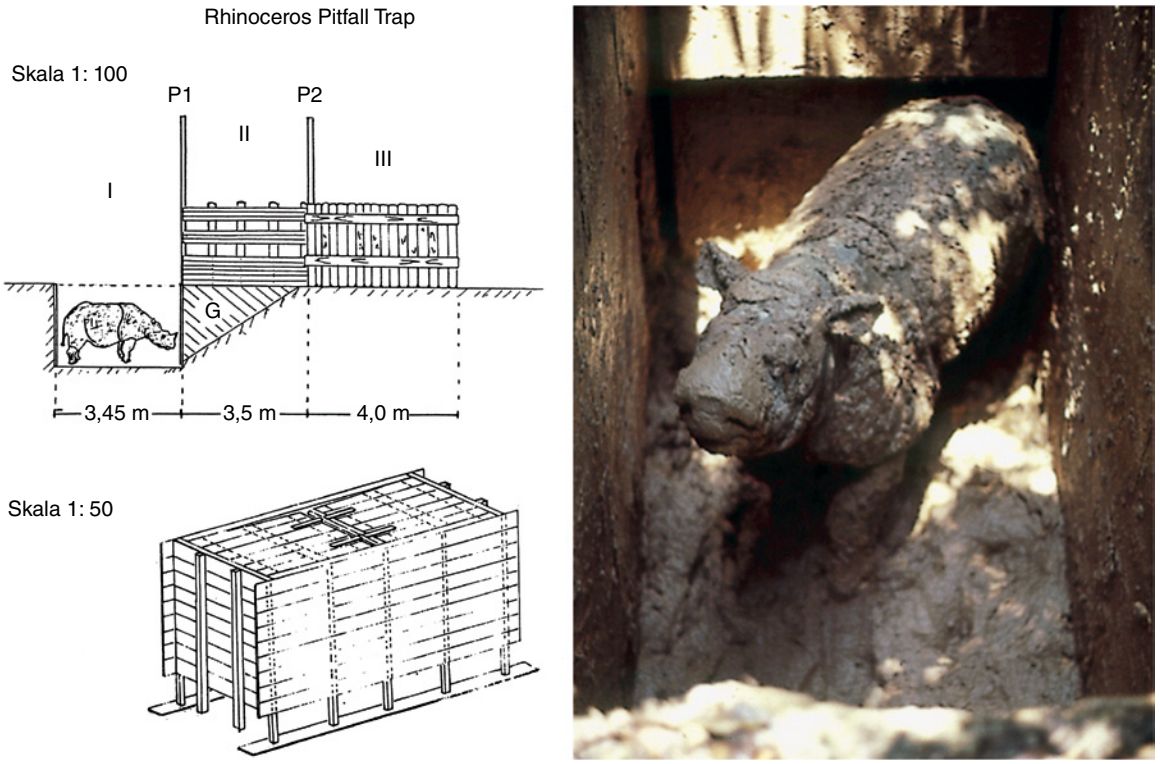

Figure 47.8 Diagrammatic sketch of pitfall capture method used in Indonesia's Riau Province for capture of wild Sumatran rhinoceros (*Dicerorhinus sumatrensis*). Sketches adapted from Sadmoko (1990), image courtesy of Mohd Khan bin Momin Khan, Malaysia Department of Wildlife and National Parks.

themselves by running into a gully or water source. With a quick induction, rhinoceroses usually go down in sternal recumbency. Occasionally the forelegs collapse first and the hindquarters remain elevated. In this situation the full weight of the abdominal organs press on the diaphragm and respiration may be compromised, especially in heavily pregnant females who have the weight of the fetus adding additional pressure. Such animals must be immediately pushed onto their sides. Most immobilized rhinoceroses will become fully recumbent, however, if the animal is still on its feet the brake rope can be placed around one of its rear legs, the blindfold over its eyes, and cotton wool in its ears.

On arrival at an immobilized rhinoceros a quick estimate of its age and body condition should be made. Older or debilitated rhinoceroses need special care. Nothing should impede respiration or push against the rhinoceros' belly, chest, or nostrils. On a slope, the rhinoceros should face uphill to alleviate pressure against the diaphragm. Field personnel must work quickly while the rhinoceros is recumbent – it helps to prepare a prioritized checklist before beginning each field capture exercise (Flamand et al. 1984; see also Box 47.1).

Box 47.1 Practical Strategies for Rhinoceros Field Anesthesia

- Darts should be tested and prepared ahead of time, loading the dart once you have visualized the rhinoceros, tailoring the dose for size, age, and condition of the animal. The rhinoceros should not be chased while the dart is being loaded. When darting from a helicopter get the dart in quickly and back off until the rhinoceros is nearing recumbency.
- Dart sites must be given special care in rhinoceroses because of the propensity for abscess formation. Rhinoceros skin is thick and tough, making drainage of subcutaneous infections unlikely without appropriate wound care. Intramammary antibiotics are often used, but in the face of growing antimicrobial resistance and a need for improved antibiotic stewardship, the authors suggest infusion of a noncytotoxic antiseptic directly into the dart wound.
- Tranquilizers are often combined with potent opioids to improve muscle relaxation in recumbent animals and to help sedate and calm the rhinoceros during transport.

- The addition of hyaluronidase, a hydrolytic enzyme that increases tissue permeability, greatly improves drug absorption and can markedly shorten the induction time.
- A lower opioid dose must be used for rhinoceroses that are in bomas, debilitated, old or where you cannot get to the immobilized animal quickly. *Be very careful with animals in poor body condition*. In most other situations underdosing of opioids is contraindicated for free-range capture of rhinoceroses.
- In general, any need for repeat darting of animals following partial or incomplete injection of immobilizing agents should redeliver the original full immobilizing dose. This is a useful rule for captive animals as well since repeat darting is often associated with excitation and prolonged drug effects if titration is attempted.
- A rapid induction shortens the period the rhinoceros is moving in a seminarcotized state and reduces injury, especially when immobilizing rhinoceroses in rough terrain. A quick induction also limits exertion and physiological stress, and associated elevations in body temperature, heart rate, and oxygen consumption.

Other Drugs and Immobilization Options

- Opiates: Carfentanil at 1–1.2 and 0.9 mg (captive white and black rhinoceros, respectively; Rogers 1993a, Portas 2004) and 3 mg for wild adult rhinoceros (Hofmeyr et al. 1975; De Vos 1978); etorphine at 1.8 mg plus 30 mg fentanyl OR fentanyl alone at 60 mg (black rhinoceros; Rogers 1993b; Kock et al. 2006); thiafentanil mixed with etorphine 2–2.5 mg thiafentanil plus 2–2.5 mg etorphine (avoid in white rhinoceros).
- Non-opiates: Medetomidine and butorphanol combinations (Morkel & Nel 2019).

Butorphanol and nalbuphine in African rhinoceros (Tables 48.2 and 48.3):

- To *improve respiration*, administer small incremental 5-mg doses of butorphanol or nalbuphine intravenously to effect (black rhinoceroses are very sensitive so be careful of sudden arousal). Butorphanol or nalbuphine may be used at 20–40 mg in a similar fashion for improving respiration in the white rhinoceros.
- To *walk a rhinoceros*, standard practice is now to give butorphanol at 10× (up to 20×) the etorphine (M99) dose (10–20 mg of butorphanol per 1 mg of M99) for white rhinoceros. Butorphanol can be incorporated into the initial dart or given at the time of recumbency or loading. Others give 1 mg of diprenorphine plus butorphanol followed by small incremental doses of butorphanol.
- For *transport*, wake white rhinoceros with 10–20 mg of butorphanol per 1 mg of M99 or give diprenorphine at 2.4× M99 dose (generally a lively wake up). Can also give 1–2 mg naltrexone with diprenorphine or later in transport to prevent pushing in crate. Can give 1–2 mg of diprenorphine intravenously if animal is pushing or collapsing in crate.

African Rhinoceros Wild Anesthetic Regimens

White Rhinoceros (*Ceratotherium simum*)

With the high doses of opioids used to speed induction under field conditions, the safe anesthesia of wild white rhinoceroses represents one of the most challenging branches of rhinoceros anesthesia (Table 47.1). Hypoxia, hypercapnia, hypertension, tachycardia, and acidosis are common physiologic abnormalities reported in anesthetized white rhinoceroses (LeBlanc et al. 1987; Heard et al. 1992; Hattingh et.al. 1994; Bush et al. 2004; Haw et al. 2014). A variety of techniques have been adopted to help alleviate the significant opioid-induced cardiopulmonary depression in African rhinoceroses. These include use of partial agonist-antagonist agents to reverse the μ-regulated opioid respiratory depression, respiratory stimulants such as doxapram, nasal or tracheal insufflation of oxygen, and incorporation of mixed agonist-antagonist agents into more potent opioid-based protocols to influence receptor effects (Kock et al. 1995; Radcliffe et al. 2000a; Bush et al. 2004, 2005; Fahlman et al. 2004; Haw et al. 2014).

Opioid doses for field anesthesia of adult white rhinoceroses range from 3 to 4.5 mg of etorphine plus 40–60 mg of azaperone or 10–20 mg of detomidine (Table 47.1; Rogers 1993a; Kock et al. 1995; Bush et al. 2004). When loading white rhinoceroses, lower azaperone doses (20–30 mg) are used to preclude recumbency in the crate (C. Moueix, pers. comm. 2019). Hyaluronidase (Hylase; 5000 IU) is often incorporated into darting protocols for rhinoceroses to shorten induction times (Morkel 1989). White rhinoceroses stopped moving 2–3 minutes sooner with hyaluronidase, but often remained standing (Kock et al. 1995). Fentanyl was once incorporated into drug cocktails for white rhinoceroses but is rarely used today (1 mg of etorphine is equipotent to 15 mg of fentanyl; Rogers 1993a). The parasympatholytic agent hyoscine was historically combined with opioids for its sedative and

amnesic properties as well as to induce "temporary blindness" by pupillary dilation (Player 1972; Rogers 1993a). However, its use is no longer widely accepted because of undesirable side effects and it is now considered obsolete (Kock et al. 1995; Raath 1999).

An extensive study of white rhinoceros anesthesia incorporating several drug protocols and 141 immobilizations over a 2-year period was conducted in Zimbabwe to enable dehorning operations (Kock et al. 1995). Initial immobilization mortality was quite high at 7% and was primarily attributed to hypoxemia and cardiovascular collapse. Subsequent captures utilized lower opioid immobilizing doses and simultaneously incorporated routine use of nalorphine (10–20 mg) or nalbuphine HCl (20–40 mg) to help improve respiration, especially in longer procedures where mortality was most prevalent. Of the various drug combinations tested (etorphine alone and in combination with fentanyl, xylazine, or detomidine), the etorphine-detomidine combination was considered superior because it was empirically judged as smoother and more rapid (no statistical significance). Pulse rates and creatinine phosphokinase (CPK) levels were significantly lower with the etorphine-detomidine combination, suggesting improved cardiac function and less muscle damage, respectively (Kock et al. 1995). Good muscle relaxation was observed without the rigidity and paddling common with use of potent opioids in the white rhinoceros. The ratio of etorphine to tranquilizer was critical and dose dependent, likely reflecting differences in drug pharmacology and onset of action.

An effective alternative for mitigating muscle rigidity in wild white rhinoceroses is the use of midazolam (Radcliffe & Morkel 2007; Table 47.1). Since immobilized white rhinoceroses are often first encountered in a standing position with a rigid body posture, IV midazolam at 5–20 mg is effective in inducing good muscle relaxation and recumbency. The Zimbabwe workers noted that even small incremental increases in etorphine in the initial immobilizing dose or redosing with etorphine resulted in poorer muscle relaxation and increased head shaking, jerking, and limb paddling (Kock et al. 1995). Midazolam has excellent muscle relaxation properties and is a useful adjunct in these situations.

Black Rhinoceros (*Diceros bicornis*)

Capture-related stress is a significant factor in field immobilization of the black rhinoceros, resulting in morbidity and mortality in the post-capture period (McCulloch & Achard 1969; Keep 1973). Rapid immobilization using high opioid doses in combination with hyaluronidase (or more potent agents such as thiofentanil alone or mixed 50:50 with etorphine) is the single most critical factor in reducing stress during black rhinoceros capture operations (Morkel 1989, 1994; Kock 1992). Furthermore, higher etorphine doses and use of hyaluronidase were associated with significantly shorter induction times, lower body temperatures, shorter distances moved, and reduced muscle damage, as evidenced by lower CPK and lactate dehydrogenase levels (Kock 1992). Although two accounts list 3 mg of etorphine as a standard opioid immobilizing dose for wild black rhinoceroses (Kock et al. 1990; Rogers 1993b), subsequent study suggests that 3 mg of etorphine is inadequate due to prolonged induction periods and associated capture stress (Kock 1992). Based on a review of published material and considerable author experience, 4 mg of etorphine is recommended as a good standard dose for an adult black rhinoceros bull or cow in good body condition (Table 47.1; Morkel 1989, 1994).

A scaled-down opioid dose should be utilized in young animals or those in poor body condition, but in all other circumstances a low dose of etorphine is contraindicated for free-range capture of the black rhinoceros (Kock et al. 2006). Azaperone is incorporated into etorphine-based African rhinoceros immobilization protocols at 40–60 mg total dose (Table 47.1). Black rhino immobilization and post crating tranquillization may be improved at higher azaperone dosages (180–200 mg in the dart) by reducing the stormy walking that typically follows partial antagonism with nalorphine or butorphanol (P. Nel, pers. comm., 2019). Concentrated (100 mg/mL) azaperone solutions should be carefully examined before use as they often crystallize under field conditions in cold temperatures. Xylazine or Detomidine (100 or 10 mg per adult, respectively) can be substituted for azaperone based on individual preference.

A disparity is evident in the opioid dose required for immobilization of the various subspecies of black rhinoceroses. The desert subspecies (*Diceros bicornis bicornis*) needs a slightly higher dose than the other subspecies. While 5 mg or even 6 mg of etorphine may be necessary for an adult *D. b. bicornis* in good condition, 4 mg is usually more than adequate for a comparable response in animals of the *D. b. minor* or *D. b. michaeli* subspecies. Not only is there variation between subspecies but there also appears to be some difference among individuals. The capture veterinarian must therefore be aware of these vagaries in dose response and be prepared to respond if an animal reacts unfavorably.

Asian Rhinoceros Wild Anesthetic Regimens

Indian or Greater One-horned Rhinoceros (*Rhinoceros unicornis*)

Techniques for field anesthesia of the greater one-horned rhinoceros were developed to meet research needs, including the elucidation of basic ecology, genetics, social

organization, and dispersal biology (Dinerstein et al. 1990; Dinerstein 2003). Furthermore, translocation programs are proving essential for reaching long-term population management goals for *Rhinoceros unicornis* in India and Nepal. Capture of wild greater one-horned rhinoceroses is usually conducted from atop trained elephants to facilitate finding and darting of rhinoceroses among the dense tall-grass habitats in the floodplain grasslands and riverine forests where these rhinoceroses flourish. In addition to providing an elevated platform, elephants (10–15 animals) are used to surround the target rhinoceros before and after darting to facilitate observation of the animal during induction and to prevent escape into open water (Dinerstein et al. 1990).

Adult greater one-horned rhinoceroses weigh an estimated 2000 kg, with males slightly larger than females. Dinerstein and colleagues immobilized 39 animals (representing 51 events) using a combination of etorphine and acepromazine (2–2.5 and 10 mg, respectively) delivered via remote IM injection either in the shoulder or rump using Cap-Chur darts with 5-cm needles (Table 47.1; Dinerstein et al. 1990). One adult female was immobilized with carfentanil (0.7 mg) and all animals were successfully reversed in the field using diprenorphine HCl (M50:50). Induction times were found to be significantly longer in breeding versus nonbreeding males, with the former group rarely moving far from the site of darting. A large disparity in induction times was noted across all age and sex groups, presumably related to variable drug delivery from dart placement among the thick skin folds characteristic of the species (Dinerstein et al. 1990).

Javan or Lesser One-horned Rhinoceros (*Rhinoceros sondaicus*)

There have been no published reports describing field capture or anesthesia of the Javan rhinoceros. As with the Sumatran rhinoceros, pitfall trap methodologies rather than stockade style traps are recommended for capture of lesser one-horned rhinoceroses in the rainforest environment provided the risks of flooding can be controlled (Nardelli 1987a; Sadmoko 1990). Field anesthesia is also possible, especially where animals are pushed out of the forest by human activities, and should be based on extrapolation of the best available information from the other Asian species.

Sumatran Rhinoceros (*Dicerorhinus sumatrensis*)

Several intensive operations have been conducted to capture wild Sumatran rhinoceroses using corral or stockade traps with little success (Abdullah 1987; Sadmoko 1990). In one instance an adult female Sumatran rhinoceros suffered severe head injuries and acute death following capture in a stockade trap from apparent panic-related self-trauma (Nardelli 1987b). Planned capture of wild Sumatran rhinoceroses in the forests of Southeast Asia has been most effective with the use of the pitfall trap. Pitfall traps (measuring 10 × 4 × 8 feet, length × width × depth) incorporate strong plywood walls to preclude landslides and a breakaway false ceiling that drops the animal into the excavated pit beneath (Fig. 47.8). Site selection favoring heavily used rhinoceros trails was considered the single most important criteria for success or failure of the pitfall trap (Abdullah 1987). Nevertheless, pitfalls suffer from significant problems. In many Sumatran rhinoceros areas poor drainage results in flooding of the pit despite careful preventive measures. Use of pumps to clear the flooded pits is recommended, but such equipment can delay capture due to the introduction of noise, smells, and other signs of human presence. Interference from nontarget species is also a common hazard; tapir, elephants, cattle, and even human beings have fallen into pitfall traps despite sign boards erected for the benefit of man (Abdullah 1987)!

Due to the dense nature of the rainforest environment and rare sightings of individual rhinoceroses therein, routine chemical capture techniques developed for Asian and African rhinoceroses are considered of high risk as an animal may be lost in the darting process. Increasingly, however, animals are being pushed from the jungle by human encroachment and once beyond the protective boundary of the forest are immediately threatened. In these circumstances, pitfall capture methods are not feasible and chemical capture techniques are indicated. Therefore, the capture process for an "at-risk" Sumatran rhinoceros found wandering within a Southeast Asian village or otherwise outside a protected area should be approached with careful planning of some urgency. Once the appropriate National Park, Rhinoceros Protection Unit (RPU), and sanctuary staff have been contacted the following stepwise approach to capture and translocation is suggested (Radcliffe et al. 2002).

Guidelines for Capture of Displaced Sumatran Rhinoceroses

Secure the Immediate Area

In the event a wild Sumatran rhinoceros is found wandering outside a protected area, the first priority should be to secure the area from villagers and would-be poachers to prevent the animal from being shot or otherwise harmed before capture or relocation of the rhinoceros is possible.

Determine Relocation Strategy

If possible, a small core group of decision makers should be formed to make immediate assessment of the risks and benefits of rhinoceros relocation. If the rhinoceros is unharmed and close to a protected area (<10 km), then it may be

desirable to move the rhinoceros without capture by pushing it back toward the forest. If the animal is injured or otherwise in need of medical attention or far (>10 km) from the forest, a decision should be made to capture the animal.

Make a Plan for Rhinoceros Capture

Considering the high risks associated with capture without using chemical restraint (i.e., the rhinoceros is physically restrained following an extensive stressful chase without the use of routine chemical capture methods), this approach is not considered safe or desirable (Fig. 47.9). The following are suggested guidelines and methodology for capture of at-risk Sumatran rhinoceroses outside a protected area.

Capture Method 1: Field Capture using Chemical Restraint

If a trained capture team is available (i.e., within 1 day's travel time) then it may be wise to have the RPU ranger staff carefully monitor and secure the rhinoceros and surrounding area from a distance without pushing the animal to run as the rangers await the capture team. A rapid induction and recumbency will be essential for safe capture of a tropical ungulate species such as the Sumatran rhinoceros that may risk drowning or suffer from capture myopathy.

Figure 47.9 Like the other rhinoceros species, the Sumatran rhinoceros (*Dicerorhinus sumatrensis*) is prone to capture myopathy. Here a wild "hairy" rhinoceros is restrained with a girth rope in hopes of moving the animal into a temporary boma. Hyperthermia is best avoided by limiting chase periods and liberal application of water. Image courtesy of Sugiyo, Wildlife Conservation Society, Indonesia Program.

For field anesthesia of the Sumatran rhinoceros, a combination of equal parts butorphanol and azaperone (80 mg each) is recommended for simplicity and its inherent safety for both rhinoceros and people alike (Table 47.1; Radcliffe et al. 2002). However, if a well-trained veterinary capture team is available then use of more potent opioids such as etorphine combined with azaperone and hyaluronidase (2 mg, 80 mg, and 5000 IU, respectively) or the newer etorphine-butorphanol-midazolam protocols may be considered depending on the situation (Bush et al. 2011). If the rhinoceros is already compromised from a chase or is restrained by a snare, the use of the safer butorphanol protocol is preferable to the potent opioids (Table 47.1). The butorphanol-azaperone combination may require confinement within a temporary boma or some additional restraint via a body or head rope to facilitate crating in healthy animals.

Capture Method 2: Field Capture by Erecting Temporary Boma

The Sumatran rhinoceros is perhaps the only species of rhinoceros that can be captured by human physical restraint alone, albeit after much chasing and associated capture stress. Therefore, if a trained capture team is not available and the rhinoceros is in immediate peril, physical capture can be a feasible option. To begin, the animal can be followed from a safe distance and without excessive chasing until the rhinoceros is located within an area where it is resting and approachable (i.e., in water or other suitable location; Fig. 47.10). Large rolls of shade cloth or tarpaulin are then carefully erected without disturbance to form a temporary boma surrounding the rhinoceros that will facilitate sedation, crating, and transport. Once the animal is restricted within the confines of the artificial boma, hand-injection or pole-syringe delivery of the butorphanol-azaperone combination will facilitate safe crating and transfer. The boma method is not likely to eliminate the long chase periods and accompanying stress, but it was effective in the recent capture and relocation of a young adult Sumatran rhinoceros in Indonesia (Fig. 47.9).

Sumatran Rhinoceros Transport

For Sumatran rhinoceroses that are conditioned to a crate prior to capture (i.e., captive animals or wild animals that allow crate training *in situ* such as in an oil palm

Figure 47.10 A wild Sumatran rhinoceros (*Dicerorhinus sumatrensis*) undergoing "hand translocation" without the use of chemical restraint after displacement from a protected forest reserve in Indonesia. Although this animal survived significant capture-related morbidity, chemical capture techniques are preferred if trained staff are readily available. Image courtesy of Chandra Putra, Way Kambas National Park, Sumatra.

plantation) there is often little need for sedation during transport. However, for animals captured in a pitfall trap, the use of both short-acting and long-acting tranquilization is indicated. In November of 2018, a Sumatran rhinoceros named Pahu was captured in a pitfall trap in East Kalimantan, Indonesia. The estimated 400-kg animal received 50 mg of zuclopenthixol acetate IM by pole-syringe shortly after capture, representing the first use of this long-acting tranquilizer in this species (Candra et al., unpubl. report). The zuclopenthixol acetate produced a calming effect during the 36 hours the animal remained in the pit while the capture team constructed a ramp and moved the transport crate into position. Two days after capture and while the animal awaited transport overland by truck, the rhinoceros became agitated in the crate and 40 mg of azaperone was given IM. The animal was calm within 20 minutes and 6 hours later the rhinoceros was loaded onto the back of a 4 × 4 pickup truck to begin transport to a nearby sanctuary. Following the 16-hour road transport the animal was given a second dose of 50 mg of zuclopenthixol acetate IM by pole-syringe immediately prior to off-loading into the sanctuary boma. The capture and translocation were a success.

Rhinoceros Crating and Transport

The moving or translocation of rhinoceroses is a specialized branch of rhinoceros anesthesia that has been practiced since the first African rhinoceroses were saved from the rising waters of Lake Kariba. The crating and relocation of rhinoceroses is now standard practice as urgent conservation measures, including enhanced animal monitoring and protected area management, have become effective tools in the fight against poaching (Hitchens et al. 1972; Flamand et al. 1984; Henwood 1989).

Walking a Rhinoceros

If a crate cannot be placed directly in front of the anesthetized animal, the rhinoceros can be "walked" a distance and guided into the crate (Fig. 47.11 and Table 47.2). When the rhinoceros becomes recumbent, the blindfold, cotton wool, head rope, and brake rope are applied. Four to six people are stationed on each rope, two people on each shoulder, one person to the side leading the team, and two people walking in front of the rhinoceros, clearing obstacles in its path. The rhinoceros is given small incremental doses of IV diprenorphine or butorphanol depending on species (Table 47.2). Butorphanol can be given IV at 10–20 times the etorphine dose to walk a white rhinoceros into a crate. Mixed opioid antagonists should be used with caution in black rhinoceroses and at lower doses with smaller increments compared to white rhinoceroses. Regardless of antidote used, a prodder is judiciously applied to the feet just above the nail, or to the muzzle or perineal area to get the rhinoceros to stand and keep it moving. After each dose, wait a few minutes (up to 10 minutes) and check the rhinoceros' response to the prodder or by squirting water in the ear. If there is no response, give another dose of the partial agonist-antagonist. Once the rhinoceros stands, it should begin to stagger forward and can then be readily guided with the head rope and by the people on the sides. If the rhinoceros moves too fast, go slowly with the head rope and pull the brake rope to slow the moving

Figure 47.11 "Walking" an etorphine-immobilized white rhinoceros (*Ceratotherium simum*) using ropes and trained personnel to guide and stabilize the narcotized animal.

rhinoceros. Particularly with young and fractious individuals it is important to slow the rhinoceros as it approaches the crate so it does not traumatize itself on entrance. A rhinoceros that charges too quickly into the crate can strike the far wall with such force to avulse the horn or crush the nasal bones. To preclude problems with loading, diazepam 10–15 mg IV 10 min before waking the rhinoceros helps to keep the animal calm when aroused in the crate.

Black Rhinoceros Crating

The recent decline in the availability and manufacture of the partial agonist-antagonist nalorphine in southern Africa has necessitated the use of alternative techniques for partial reversal and crating of black rhinoceroses in the field. Initial trials using similar agents such as nalbuphine and butorphanol have worked (sometimes quite well) although they have also been associated with irregular outcomes and responses in the black rhinoceros. Nalbuphine provides a satisfactory partial reversal that facilitates crating of black rhinoceroses, but appears to predispose crated animals to dog-sitting and squatting, which can lead to serious myopathy and inability to stand (Fig. 47.12). Butorphanol given in higher doses (25–30 mg for an adult animal) also provides a reliable partial reversal for crating of black rhinoceroses. However, such animals seem prone to head pressing in the crate and require constant supervision. Head pressing in the black rhinoceros seems to be correlated with too much opioid activity; this can be countered by using naloxone (one or two 0.4-mg ampoules) to lighten the opioid effect with a pure antagonist (P. Nel, pers. comm., 2019). If the rhinoceros is slightly too alert, one can manage the animal with azaperone, which does not cause head pressing.

Figure 47.12 Post-translocation myopathy in a black rhinoceros. Capture complications are more prevalent in animals that experience excessive chase periods, hyperthermia, or struggle to stand on crating. Image courtesy of Birgit Kötting, Etosha Ecological Institute, Namibia.

A novel approach to crating the black rhinoceros currently practiced in Namibia combines diprenorphine together with methods to limit noise and visual stimulus during transport. A muslin (mutton) cloth works well for both white and black rhinoceroses (Fig. 47.4). Animals remain remarkably tranquil, although some degree of chemical tranquilization is still necessary. The beauty of blindfolding is that a physical rather than a chemical means is used to calm the animal. The cloth must be placed with great care to avoid damage to the eyes or loss of the blindfold during crate transport. One should brush or blow any dirt from around the eyes and flush the eyes with saline, if necessary. Tying the blindfold properly takes two people – the secret is to start with a 4-m length of muslin cloth and place the middle point on the forehead directly behind the back horn. Both ends of the cloth are pulled tightly on either side of the head, making sure both eyes are closed in a normal manner. The cloth is wrapped under the jaw around the opposite side behind the back horn where it is tied securely. While wrapping, the cloth should be spread to cover the eye properly and hooked behind the jaw. Three heavy-duty cable ties 40 cm in length are used to secure the blindfold to the rhinoceros. Two ties are threaded through holes in the cloth fashioned above and forward of the eye and closed to form a loop. A third cable tie is threaded through the two loops in front of the back horn and pulled tight. The cable ties serve to pull the cloth forward securely over the eyes. If the cloth becomes loose during the journey, simply pull on a cable tie to tighten the blindfold.

Reversal and crating are then routine: a heavy hemp rope is secured around the head with a blindfold and threaded through the front of the crate. This rope will provide the forward pull on standing and will direct the rhinoceros into the forward part of the crate. A second rope is secured to one rear leg and is used as a break rope to simultaneously slow the momentum of the rhinoceros so it does not collide with the front of the crate, where horn or nasal trauma may result. A relatively high dose of diprenorphine (black rhinoceros adult 1.5–2 mg; subadult 1 mg) is given intravenously after positioning immediately in front of the crate door. The team remains quiet and the animal is not disturbed for 2 minutes. On standing, the rhinoceros is pulled forward into the crate while the break rope on the back leg slows the rhinoceros. Once inside the crate, the rhinoceros is secured by sliding three pipes into place at the rear of the crate and the rear doors are closed. This protocol consistently produces a lively and relatively awake

rhinoceros inside the crate that remains calm because of the combined use of a secure blindfold and earplugs.

Rhinoceroses blindfolded with muslin cloth travel well and can be given other sedatives, including azaperone (60–100 mg) or additional opioids (etorphine or butorphanol), during transport as needed. From about 4 hours onward an additional 0.05–0.1 mg of etorphine (usually with about 60 mg of azaperone) is administered, and can be repeated every 2 hours. The veterinarian (or someone with a high level of experience with opiates) must remain with the animal for the entire trip to evaluate and top-up as needed. Straightening with a prodder applied lightly to the forehead, neck or shoulder is often necessary in the first few hours. The muslin cloth can be kept on for as much as 36 hours. It is essential to put the cloth on tightly, make sure it is 100% clean, and ensure that no sand or dirt gets into the rhinoceros' eyes.

The beauty of the tight blindfold and blocked ears is that you can wake up the rhinoceros to a large degree (and therefore prevent pushing), but because the animal cannot see or hear it is very unresponsive and rarely gets excited – the effect is quite remarkable. However, the muslin cloth blindfold is inadequate by itself for crate transport and additional tranquilization using repeated ultra-low doses of etorphine and azaperone is essential along the road.

Tranquilization During Transport

All black rhinoceroses require tranquilization during transport (even most crate-conditioned animals) to preclude excessive struggle and associated trauma (Table 47.2). Other rhinoceros species tolerate transport better than black rhinoceroses, but still often benefit from some sedation. The veterinarian must always travel with the rhinoceros and be prepared to give additional sedatives or even narcotics if needed. It is imperative that the veterinarian anticipates the animal's tranquilization needs as waiting until the rhinoceros is alert and bouncing around will risk unnecessary trauma to both animal and attendant. Additionally, a cool animal is generally more relaxed than an overheated one.

Rhinoceroses settle into the rhythm of transport quite well after just a few hours. However, as the short-acting tranquilizers begin to wear off, the animal may become excited if suddenly startled (i.e., from stopping, off-loading, etc.). The rhinoceros can be redosed with tranquilizers while the vehicle is in motion or stop, inject, and start moving again immediately. In most instances, hand-injection is the best method to deliver additional tranquilizer. A 20-gauge, 1.5-inch needle is inserted into the lateral muscles of the neck while avoiding the nuchal region. Beware of the head and horn during neck injections. For restless individuals, the gluteal region also works well. Once the rhinoceros has settled, attach the syringe and inject the drug. A pole syringe can also be used, but hand-injection is preferred because it precludes the startled response resulting from the jab of the pole. Beware of coring, where the rhinoceros' skin may block the needle lumen. An IM injection takes 5–10 minutes for first effect; for a faster response, an IV injection into the ear vein is sometimes possible, although care must be taken to avoid the dangerous area around the animal's head and horn. Recumbency during transport can be beneficial or a potential problem in a crated rhinoceros, depending on the animal's position and duration of recumbency. If the rhinoceros lies down while the vehicle is moving, the rocking and bouncing action of the truck helps to facilitate limb circulation. Beware, however, if the rhinoceros lies down for a long period (>45 minutes) since the complications from compartment syndrome are serious. Rhinoceroses heavily sedated with opioids often struggle to work out a way to lie down; however, if they manage to do it once, they will lie down more easily thereafter.

Short-acting tranquilizers such as azaperone, xylazine or detomidine, and diazepam or midazolam are useful agents to produce a calming effect in rhinoceroses during transport. Azaperone is the tranquilizing agent of choice and can be repeated every 4–6 hours as needed (Rogers 1993a,b; Kock et al. 2006). A 40 mg/mL azaperone solution is a convenient preparation and mixes well with etorphine for IM administration to a fractious, crated rhinoceros. The administration of opioids, either alone or in combination with IM azaperone or diazepam, is the only effective way to preclude an excited black rhinoceros from traumatizing itself inside a crate (Table 47.2). Etorphine and azaperone (0.05–0.15 and 100–200 mg, respectively) are delivered by hand-injection or pole-syringe with sedation achieved in 5–10 minutes for durations of 2 hours or more.

Long-acting tranquilizers can help to calm an animal, but are inadequate by themselves to sedate an excited animal during transport. Zuclopenthixol acetate (Clopixol Acuphase, 25–150 mg per adult rhinoceros) takes about an hour to provide sedative effects after administration, while perphenazine enanthate (Trilafon, 200–400 mg per adult) takes about 12 hours for the first noticeable effects to appear (Swan 1993; Kock et al. 2006; Table 47.2). Perphenazine works well for the translocation of black rhinoceroses while caution should be exercised in white rhinoceroses as its use has been implicated in anorexia (Portas 2004; Kock et al. 2006). Higher doses of long-acting tranquilizers should be avoided before release from the crate or boma since it may cause animals to lie down, creating a hazard in hot environments or where predators are present.

Black Rhinoceros Off-loading from Crate

Black rhinoceroses, especially juveniles and subadults, are notorious for being aggressive and sometimes even self-destructive to themselves and the crating equipment (trucks, crates, etc.) at off-loading sites. In Namibia, where rhinoceroses are often moved from veldt to veldt without the use of bomas and adaptation periods, the capture team uses a technique for off-loading that is effective and largely eliminates the aggressive phase. At the off-loading site, the rhinoceros is resedated inside the crate using a low dose of etorphine (adult 0.1–0.2 mg) combined with azaperone (adult 80–120 mg) and allowed to narcotize over a period of several minutes. Once the rhinoceros is head pressing or otherwise sedated, the muslin blindfold and earplugs are removed and the etorphine antagonist administered. A standard dose of 12–18 mg of diprenorphine for an adult animal is administered by IM (not IV) injection and the crate doors are opened. As the rhinoceros regains its first levels of awareness it walks slowly from the crate even while still sedated and partially narcotized. These black rhinoceroses tend to walk directly away from the crate and into the veldt without the characteristic aggression and attack of the crate or related equipment. As the animal continues to walk away from the off-loading site it becomes more fully aware of its surroundings and ambulates from the site in a normal fashion. This protocol has largely eliminated the aggression and self-trauma that is often characteristic of black rhinoceroses at off-loading.

Alternative Rhinoceros Anesthesia Techniques

Antidote Choice

Following IV antidote administration, a recumbent rhinoceros will stand within 60–80 seconds. Response to the antidote is first noted as an increase in the depth and rate of respiration and movement of the ears and eyes. Rhinoceroses get to their feet quickly and are immediately strong and aggressive. A rhinoceros should always be moved into sternal recumbency before giving the antidote or it may bash its head on the ground as it attempts to rise from the lateral position. IM dosing of the antidote is often preferred for arousal of rhinoceros cow–calf combinations so the pair awake slowly and have time to join together without dashing off in opposite directions. If IV dosing is desired with recovery of cow–calf pairs, the cow is injected first, 30 seconds before the calf.

Out of tradition, opioid antagonists are dosed using empirically derived ratios rather than on a milligram per kilogram basis; for the pure opioid antagonist, naltrexone, dosage ratios of 20–50 times the etorphine milligram dose and 90–100 times the carfentanil milligram dose are considered standard for captive rhinoceroses (Swan 1993; Allen 1996; Kock et al. 2006). Renarcotization has been reported in the white rhinoceros, but it is a rare occurrence in the black rhinoceros (Kock et al. 1990; Portas 2004). Field workers frequently use lower naltrexone doses (12.5:1 naltrexone to etorphine ratio) without a problem (Kock et al. 1995), but sedative signs at these doses have been reported in white rhinoceroses and a minimum of 40:1 is therefore recommended to preclude renarcotization (Rogers 1993a; Kock et al. 1995; Portas 2004). While naltrexone is considered the agent of choice for complete reversal of narcotic anesthesia, a number of scenarios arise under both captive and field conditions where a full reversal of an opioid-based procedure is undesirable.

The choice of antagonist and its desired action is dependent on two factors: species and location. Black rhinoceroses are reversed to load into a crate with nalbuphine or butorphanol (Kock et al. 2006; Radcliffe & Morkel 2007; Tables 47.1 and 47.2). In the boma, *Diceros bicornis* are reversed with naltrexone, although very nervous or aggressive individuals may benefit from reversal with diprenorphine for its sedative properties. Naltrexone is used for complete antagonism in the field.

In marked contrast to black rhinoceroses, white rhinoceroses are reversed into a crate using diprenorphine with perhaps 1 or 2 mg of naltrexone. In the boma and in the field, *Ceratotherium simum* are similarly reversed with naltrexone. Diprenorphine is often used for translocation of white rhinoceroses since its partial agonist-antagonist actions provide significant narcosis during travel. However, diprenorphine has minimal agonist effects in *Diceros bicornis* and therefore should be used judiciously for transport in this species. For any partial antagonism in a crate situation it is critical that the rhinoceros be monitored very carefully to prevent excessive head pressing and occlusion of the airway or damage to the neck and limbs. A cattle prodder is a vital piece of equipment in managing sedated rhinoceroses during travel.

Rhinoceros Anesthesia Complications

With opioid-induced cardiopulmonary compromise common in anesthetized rhinoceroses, the need may arise to assist respiration. For emergency respiratory support for apnea (after having reversed the opioid with pure opioid antagonist), the animal is first pushed onto its side. A large person forces the knee and lower leg (with foot placed firmly on the ground) into the abdomen to vigorously force the abdomen diagonally upward and forward against the

diaphragm. This moves the diaphragm, forcing air into and out of the lungs and keeps the animal alive while the IV opioid antagonist takes effect. When one leg is tired use the other leg and recruit additional people to assist. Jumping on the ribs or back of the rhinoceros is ineffective and does nothing but fracture ribs and inflict unnecessary trauma. Mechanical positive pressure ventilation can be used, but few field practitioners have such equipment routinely at hand (Pohlin et al. 2019).

Myopathies are common in rhinoceroses that experience excessive chase periods or hyperthermia during capture. An especially critical period occurs at the time of crate loading and initial transport during field translocation of rhinoceroses. If stimulated to rise too early after partial reversal, animals may enter the crate and assume a rigid, semi-squatting position with their hind legs. This is undesirable and must be resolved quickly before the muscles are irreversibly damaged (Fig. 47.12). Use of the electric prodder on the head can often stimulate the animal to rise and stand, but avoid prodding the hindquarters as this can exacerbate the problem. If this does not work consider prompt IV administration of diprenorphine. A sling can also be placed under the belly of the animal, just in front of the rear legs to lift the hindquarters (using the crane on the recovery truck) until the strength has returned to the hind limbs.

A very small percentage of black rhinoceroses develop an adverse reaction that the author refers to as the "fat nose syndrome" (Radcliffe & Morkel 2007). Essentially the nostrils close up and appear edematous with a much-reduced opening to the nares. The anesthetist is often forced to hold open or pull the nostrils apart. This unusual response may indicate a hypersensitivity (anaphylactic) reaction; morphine is known to cause histamine release in humans and perhaps etorphine – derived from the same group of opium alkaloids – can produce the same uncommon effect in susceptible rhinoceroses.

New Field Anesthesia Techniques

Today's understanding of Rhinocerotidae anesthesia is truly the embodiment of many courageous pioneers who have led by exciting experimentation and hard-won experience (Harthoorn & Lock 1960; Player 1972; Young 1973; Kock et al. 2006). Yet with the immense challenges inherent in practical anesthesia of these complex mammals, innovative procedures are welcome. The newest ideas for rhinoceros anesthesia are arising from a combination of practical experience and a desire to explore the depths of pharmacology. Nowhere are such explorations more exciting than the emerging science of mixed opioid receptor action on central nervous system activity (Chindalore et al. 2005). Various opioid receptor affinities and their pharmacologic actions are well described in humans but remain little understood in animals, including the rhinoceros, which is certain to be unique in many respects. Indeed, the most exciting of these novel investigations is, at least for rhinoceros capture specialists, the incorporation of mixed agonist-antagonist opioid cocktails as part of routine field capture methodologies for the African rhinoceros (Bush et al. 2005).

Recent work by Bush and colleagues in white rhinoceroses combines a mixture of concentrated butorphanol (40–90 mg, 50 mg/mL solution) with etorphine and midazolam (2–3.5 and 25–50 mg, respectively; Table 47.1) (Bush et al. 2005, 2011). The addition of butorphanol to the anesthetic combination of etorphine and midazolam produces enhanced muscle relaxation with improved physiological parameters compared to the standard protocol of etorphine and azaperone in the white rhinoceros. Butorphanol is a mixed opioid agonist-antagonist; its agonist κ receptor produces analgesia and marked sedation while the high affinity at the μ receptor displaces etorphine and acts as a partial agonist, thereby producing catatonic effects while simultaneously reducing respiratory compromise, tremors, and rigidity. Etorphine is a μ agonist causing respiratory compromise, tremors, and muscle rigidity – these adverse μ agonist actions are partially antagonized by butorphanol and significantly reduce tremors and the cardiopulmonary depression typical of the pure opioids alone. In white rhinoceroses, practitioners now routinely give 40–60 mg butorphanol as soon as they reach the animal to improve respiratory function (C. Moueix, pers. comm., 2019). In black rhinoceroses, the butorphanol antagonism of μ-opiate actions will result in a lively rhinoceros not suitable for handling without additional sedation.

Etorphine produces an increase in sympathetic activation that produces profound impacts on cardiopulmonary physiology, including elevations in heart rate and cardiac output; these effects are partially antagonized by butorphanol (Boesch et al. 2018). Following butorphanol administration blood gas values reveal a more normal pH and partial pressure of arterial carbon dioxide ($PaCO_2$) while blood pressures remain lower than with the standard pure opioid agonist protocols. Administering diprenorphine, a μ antagonist, intravenously 12 minutes into the anesthetic episode reverses etorphine but not butorphanol, further counteracting adverse μ effects of etorphine while preserving butorphanol sedation effects, therefore if inadvertent opioid overdosage should occur, compromised physiological parameters can be rapidly corrected without losing control of the animal. These discoveries may help to bring field rhinoceros capture into the realm of safety

realized with captive animals where butorphanol-based protocols are now standard replacements for more potent opioids (Radcliffe et al. 2000a; Portas 2004).

Suspension of Black Rhinoceroses for Air Lifting

Aerial translocation of captured black rhinoceroses living in rugged environments in Namibia has been accomplished by suspending them by their feet (Fig. 47.13). To study black rhinoceroses in the hanging posture, a preliminary study was conducted by slinging immobilized animals under a crane to simulate aerial transport (Radcliffe et al. 2020). It was anticipated that suspension would compromise respiratory gas exchange more than lateral recumbency. Twelve wild black rhinoceroses were immobilized with etorphine and azaperone; half were positioned in lateral recumbency first and then suspended by their feet from a crane second, while an equal number were suspended first and then moved into lateral recumbency. All animals were hypoxemic and hypercapnic in both postures. When suspended by the feet, PaO_2 and partial pressure of alveolar oxygen (PAO_2) were 4 mmHg greater and $PaCO_2$ was 3 mmHg less than in lateral recumbency. Tidal volume and minute ventilation were similar between postures.

Although suspension by the feet for 10 minutes impaired pulmonary function to a similar degree as that imposed by lateral recumbency, PaO_2 was higher and $PaCO_2$ lower during suspension than during lateral recumbency. These findings suggest that airlifting immobilized black rhinoceroses compromised their cardiopulmonary system to a similar degree as lying in lateral recumbency. Arterial blood gas partial pressures might be marginally improved in the hanging posture, but the profound hypoxemia and hypercapnia remains clinically important.

Rhinoceros Calf Anesthesia

Captive Calf Protocols

Anesthesia of captive white and black rhinoceros calves is safely accomplished with butorphanol alone or in combination with detomidine (Radcliffe et al. 2000c; Langan et al. 2001; Gandolf et al. 2006). Due to high sensitivity to opioid agents, rhinoceros calves respond very well to sedation and anesthesia with mixed agonist-antagonists, precluding many of the adverse cardiopulmonary depressant effects observed with more potent pure agonists of this class. Furthermore, a rapid onset of action is attained by IV delivery or a slower induction by IM administration, with both methods proving safe and effective for serial anesthesia (Gandolf et al. 2006; Table 47.5). The combination of the alpha-2 agonist detomidine along with butorphanol was thought to enhance muscle relaxation and depth of anesthesia with IM use in white rhinoceros calves. Complete reversal is achieved using naltrexone at four to five times the butorphanol milligram dose and yohimbine HCl (Yobine) or atipamezole at 0.125 mg/kg for antagonism of the alpha-2 agent.

Cow and Calf Field Capture

Field immobilization of juvenile rhinoceroses is not without inherent risk as calves may separate from their dams after darting or become recumbent at different times despite

Figure 47.13 Hanging posture of a black rhinoceros (*Diceros bicornis*) using a crane on a flatbed truck to simulate aerial suspension (left). Helicopter-assisted aerial slinging of a black rhinoceros by its feet during translocation operations in northwest Namibia (right).

Figure 47.14 Anesthesia of rhinoceros calves is challenging, particularly under field conditions, where darting of the cow–calf pair must be well coordinated to limit stress on both parent and offspring.

concurrent drug delivery (Fig. 47.14). Additionally, calves are more susceptible to capture stress, hyperthermia, and post-capture morbidity and mortality in boma situations (Kock et al. 1995). Translocation of cows with calves less than 18 months of age can be traumatic and is best avoided while movement of very young calves 2–3 months old is particularly high risk. Even with successful translocation, it can be difficult to reunite the cow and calf as the stress of capture and confinement often results in adult aggression directed toward the calf or the cow drying-up. Methods for opioid sedation (0.2 and 0.05 mg of etorphine for a cow and calf, respectively) have been used to facilitate boma reintroduction of cow–calf combinations (Kock et al. 2006). The wild black rhinoceros cow is solitary by nature and usually retreats to a quiet spot to calve and will stay there for the first month afterwards. Therefore, if a black rhinoceros gives birth in a boma she rarely manages to raise the calf.

Opioid doses lower than those reported for adult animals are utilized for juvenile rhinoceroses, with subadults receiving approximately one-half the adult dose. For example, when combined with a tranquilizer subadult African rhinoceros (age ~2.5 years) should receive 1.75–2 mg of etorphine while very young calves (age 2–3 months) can be immobilized with as little as 0.5–1 mg of etorphine (Rogers 1993a,b; Morkel & Nel 2019; Table 47.5). A marked difference is observed in the escape behavior of African rhinoceros cow–calf pairs and should be anticipated during the chase and capture. White rhinoceros calves run ahead of their mothers, while black rhinoceros calves run close at their mothers' heels (Kock et al. 2006).

When darting a cow with a calf from a helicopter, a fixed-wing aircraft is desirable to circle the capture site to assist with spotting. As a general rule, the cow is darted first and about a minute later the calf is darted (Kock et al. 2006). If the timing and darting are good, the pair will often go down together. Should the pair split up, the fixed-wing aircraft can stay with one animal. In open country where visibility is good, the calf can be darted once the cow shows early signs of narcosis. In more thickly vegetated country where it is difficult to observe two separated animals, it is better to wait until the cow shows marked effects or is even recumbent before darting the calf. If the calf splits from its mother, the position of the immobilized mother can be taken by GPS or marked with a smoke grenade or toilet paper and the calf followed. Losing sight of a darted rhinoceros must be avoided and it is therefore mandatory to have experienced trackers as part of the ground team. When darting a cow–calf pair on foot, the calf will usually stay close to its immobilized mother. If approached carefully, the calf can be darted and will generally become recumbent close to its mother; note that black rhinoceros calves are skittish and run off more easily than white rhinoceros calves.

Wild subadult greater one-horned rhinoceroses have been immobilized using the same dosage as adult animals (2–2.5 mg of etorphine plus 10 mg of acepromazine; Dinerstein et al. 1990). However, subadult animals proved more difficult to capture and often evaded darting attempts by outrunning the trained elephants that are commonly utilized for field immobilization of greater one-horned rhinoceroses in the tall grassland habitats of India and Nepal. Rhinoceros calves were immobilized with 0.5–1 mg of etorphine and 5 mg of acepromazine using shorter 2.5-cm Cap-Chur needles. As with capture of African rhinoceros cow–calf pairs, it is recommended that greater one-horned cows be immobilized before their calves. Calves did not run away and were easier to capture if the mother was immobilized first to avoid trampling risk to calves or aggression toward the ground crew (Dinerstein et al. 1990).

Conclusion

During the Indian Mutiny a British soldier fired a bullet into the regiment's cherished mascot, a rhinoceros. In a spirit of scientific inquiry the soldier tested the long-held belief – a conviction still strongly held by many since Dürer's famous rhinoceros – that its skin was held together with rivets like a knights armor and impenetrable to any volley man could throw its way. To the surprise of royalty and commoners alike the rhinoceros quickly expired.

The future of the world's rhinoceroses will remain tenuous as human conflicts over shared resources escalate

and rhinoceros horn continues to be cherished by traditional Asian societies for supposed unicorn-like mythical properties. Nevertheless, it is comforting to know that man – while solely responsible for the current crisis – is also simultaneously making strides to save the relic rhinocerotoids from their greatest enemy, ourselves. Safe anesthesia of wild and captive rhinoceroses alike will help scientists realize these conservation goals. Let us not make the same mistake as the British soldier and believe, naïvely, that the armored rhinoceros is invincible to the actions of our kind.

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