2018

EAZA Best Practice Guidelines for the white rhinoceros

(*Ceratotherium Simum*)

First Edition, July 2018 | Lars Versteege

EAZA Best Practice Guidelines for the white rhinoceros

(Ceratotherium simum)

Editor: Lars Versteege

Contact information: Safaripark Beekse Bergen,
Beekse Bergen 31, 5081 NJ Hilvarenbeek, The Netherlands

Name of TAG: Rhino TAG

TAG Chair: Friederike von Houwald (Zoo Basel, Switzerland)

Edition: 1

July 2018

EAZA Best Practice Guidelines Disclaimer

Preamble

Right from the very beginning it has been the concern of EAZA and the EEPs to encourage and promote the highest possible standards for husbandry of zoo and aquarium animals. For this reason, quite early on, EAZA developed the "Minimum Standards for the Accommodation and Care of Animals in Zoos and Aquaria". These standards lay down general principles of animal keeping, to which the members of EAZA feel themselves committed. Above and beyond this, some countries have defined regulatory minimum standards for the keeping of individual species regarding the size and furnishings of enclosures etc., which, according to the opinion of authors, should definitely be fulfilled before allowing such animals to be kept within the area of the jurisdiction of those countries. These minimum standards are intended to determine the borderline of acceptable animal welfare. It is not permitted to fall short of these standards. How difficult it is to determine the standards, however, can be seen in the fact that minimum standards vary from country to country. Above and beyond this, specialists of the EEPs and TAGs have undertaken the considerable task of laying down guidelines for keeping individual animal species. Whilst some aspects of husbandry reported in the guidelines will define minimum standards, in general, these guidelines are not to be understood as minimum requirements; they represent best practice. As such the EAZA Best Practice Guidelines for keeping animals intend rather to describe the desirable design of enclosures and prerequisites for animal keeping that are, according to the present state of knowledge, considered as being optimal for each species. They intend above all to indicate how enclosures should be designed and what conditions should be fulfilled for the optimal care of individual species.

Citation

This publication should be cited as follows:
EAZA White rhino EEP Best Practice Guidelines; L. Versteege; Safaripark Beekse Bergen; July 2018

Summary

The best practice guidelines, designed as reference manual for any professional working with white rhino are divided into two sections. Section one describes the natural biology of the white rhino whereas section two describes the best practice husbandry in captive settings. Chapters include enclosure design, nutrition, social structure, breeding, handling and veterinary care.

Contents

Contents

Contents

List of figures

List of tables

1. Introduction

One of the main goals of modern zoos worldwide is conservation (WAZA, 2018). According to the IUCN red list 37% of all evaluated species are threatened (17.291 threatened species) (IUCN, 2010a). To keep the white rhinoceros (*Ceratotherium simum*) from extinction and to maintain a healthy, sustainable captive population, the European Association of Zoos and Aquaria (EAZA) started an European Endangered Species Program (EEP) (Versteege, 2010a; EAZA, 2018). The numbers of white rhinos are increasing and therefore the white rhino is stated as near threatened since 2002 by the IUCN (IUCN, 2010b). Nowadays 654 white rhinos in captivity are registered on Species 360 worldwide, with 286 of them in European zoos. (Species 360, 2018) The first registration of a white rhino kept in an European zoo was in 1950 (Versteege, 2010a).

In this report the best practice guidelines for the white rhino (*Ceratotherium simum*) are discussed. The best practice guidelines are developed in order to help optimise the conditions for the wellbeing of the animals kept in captivity.
The guideline is divided into two sections. The first section provides general information on the species biology, conservation status, ecology, diet, reproduction and behaviour. The data is obtained from different literature sources, such as books, articles and the Internet. The second section contains information on the actual management in captivity. This section presents recommendations on the enclosure, diet, social structure, breeding, behavioural enrichment, handling and veterinary considerations. The data to write this section is acquired from different literature sources and from the results of a survey that was conducted among EAZA zoos that participate in the EEP program.

Section 1 - Biology and field data

1.1 Biology

This chapter covers basic biological information relevant to in situ and ex situ white rhinos. The taxonomy, morphology, physiology and longevity are successively discussed.

1.1.1 Taxonomy

The taxonomic position of the white rhinoceros (*Ceratotherium simum*) is described by Burchell (1817), including all living subspecies.

Kingdom: *Animalia* (Animals)
Phylum: *Chordata* (Chordates)
Sub phylum: *Vertebrata* (Vertebrates)
Class: *Mammalia* (Mammals)
Order: *Perissodactyla* (Odd-toed ungulates)
Family: *Rhinocerotidae* (Rhinoceros)
Genus: *Ceratotherium* (White rhinoceros)
Species: *Ceratotherium simum* (White rhinoceros)
Sub species: *Ceratotherium simum cottoni* (Northern white rhinoceros)
Ceratotherium simum simum (Southern white rhinoceros)

There are many common names for the white rhinoceros, including:

- White rhinoceros
- White rhino
- African white rhinoceros
- Square-lipped rhinoceros
- Square-mouth rhinoceros
- Grass rhinoceros (Kingdon, 1997)
- Burchell's rhinoceros (Rookmaaker, 2003)

The scientific name for the white rhino is *Ceratotherium simum*, in which the Greek *cerato* means 'horn' and *thorium* means 'wild beast'. The Greek *simus* means 'flat nosed' (RRC, 2018).
Ten different theories are listed to explain the name 'white rhinoceros' for an animal that is grey, not white. The popular explanation is that 'white' is derived from the African words 'wyd', 'wyt', 'weit' or 'weid' (all meaning wide), referring to the wide mouth, but this is examined and found to be unsubstantiated and historically incorrect. (Rookmaaker, 2003)

1.1.2 Morphology

Rhinos are grey and almost hairless (hairs only on ears, tail tips and eyelashes). The head hangs down and only looks up when alarmed (see figure 2). White rhinos have a wide upper lip and a noticeable hump on the back of their neck (see figure 1 and 2). The front and back feet each have three toes and the front a soft and elastic sole. (RRC, 2018; Fouraker and Wagener, 1996; Tomasova, 2006) The measurements of these mega herbivores are shown in table 1 (Foster, 1960; Pedersen, 2009; Tomasova, 2006).

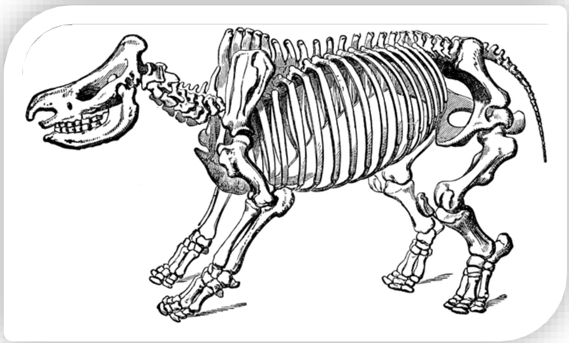

Figure 1. Illustration of the skeleton of a rhino

Table 1. ***Measurements of the white rhinoceros***

Measurements	Adult male	Adult female	New-borns
Weight	1800 - 2500 kg	1600 - 2000 kg	40 - 60 kg
Head body length	3.8 - 5 m		-
Tail length	50 - 70 cm		-
Shoulder height	1.5 - 1.8 m		-
Anterior horn	94 - 102 cm		-
Posterior horn	Up to 55 cm		-

In table 2 the dental formula of white rhinos is described. "The deciduous premolars 2, 3 and 4 are replaced by permanent premolars, while premolar 1 is not replaced" (Hillman-Smith *et al.*, 1986). Hillman-Smith *et al.* (1986) found no signs of incisors or canines in their study.

Table 2. ***The typical dental formula for white rhinos***

	Incisors	Canines	Premolars	Molars
Deciduous	0/0	0/0	4/4	0/0
Permanent	0/0	0/0	3/3	3/3

1.1.3 Physiology

Information on heart rate, respiration rate and rectal temperature is listed in table 3 (Citino and Bush, 2007).

Table 3. ***Physiologic parameter of the white rhino***

Physiologic parameter	Mean	Min.	Max.
Heart rate (beats/min)	39	32	42
Respiratory rate (breaths/min)	19	16	23
Rectal temperature (°C)	36.8	36.6	37.2

White rhinos have a very powerful olfactory sense (Pedersen, 2009; Tomasova, 2006: Grün, 2006). Hearing is sensitive when not disrupted by other environmental noises (Pedersen, 2009; Tomasova, 2006). The eyesight is poor, they can only see motionless forms between 15 to 25 meters away (Owen-Smith, 1973; Tomasova, 2006).

1.1.4 Longevity

In the wild a white rhino can reach an age of 40 to 50 years (RRC, 2018). In captivity a white rhino can reach an age of 50 years (Tomasova, 2006).

1.2 Field data

This chapter relates specifically to white rhinoceroses in the wild. It includes information on geography and ecology, diet, reproduction and behaviour.

1.2.1 Geography and Ecology

Distribution

The Southern white rhino is now the most numerous of the rhino taxonomical group. South Africa is the stronghold for this subspecies with sizeable populations in the Kruger national park and Hluhluwe-Imfolozi. Smaller populations also occur in numerous state protected areas and private reserves (some of which are also well protected). There are smaller reintroduced populations within the historical range of the species in Namibia, Botswana, Zimbabwe and Swaziland, while a small population survives in Mozambique. Populations also have been introduced outside of the former range of the species i.e. to Kenya, Uganda (meaning that the species has been reintroduced to this country) and to Zambia, as can be seen in figure 3 (Emslie and Brooks 1999). The majority (98.8%) of white rhino occurs in just four countries, namely South Africa, Namibia, Zimbabwe and Kenya (Milliken *et al.,* 2009).

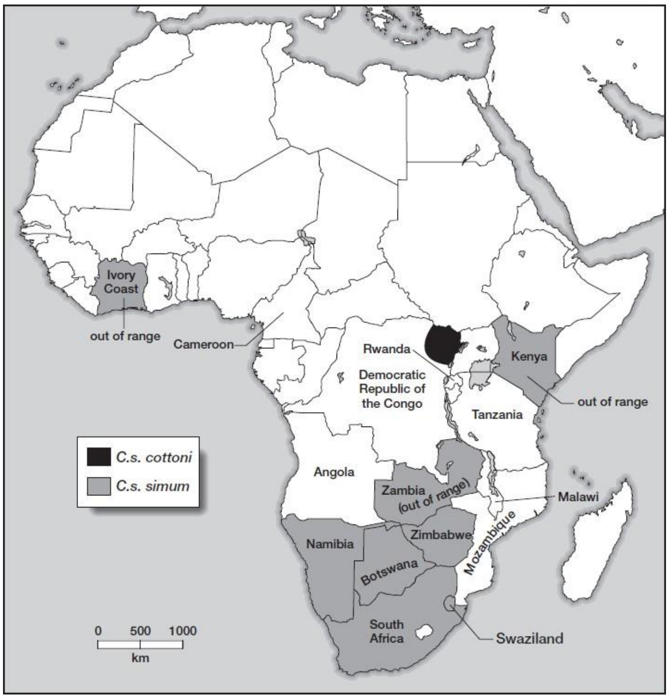

*Figure 3. Distribution of the two sub species of white rhinoceros, C.s. cottoni and C.s. simum**

* Note: At the request of certain members, the African Rhino Specialist Group (AfRSG) has a policy of not releasing detailed information on the whereabouts of all rhino populations for security reasons. For this reason, only whole countries are shaded on the map.

Habitat

White rhinos prefer short-grassed savannah with access to thick bush cover for shade and water holes for drinking as well as wallowing. The optimal habitat is a combination of grassland and open woodland. (Tomasova, 2006)

Population

On December 2015, there were an estimated 20.378 white rhinos living in the wild (see Table 4) (Milliken *et al.,* 2017). In July 2018 there were an estimated 654 living in captivity. (Species 360)

Table 4. Estimated white rhino numbers in wild- / national parks in December 2015 by country

White Rhinoceros			
	C.s. cottoni	C.s. simum	Trend since 2005
Botswana		239	up
Kenya	2	441	up
Mozambique		29	up
Namibia		822	up

South Africa		18.413	up
Swaziland		76	down
Uganda		15	up
Zambia		10	stable
Zimbabwe		330	up
Total	2	20.378	up

By the end of 2015 the numbers of white rhino in South Africa have decreased from 20.604 at the end of 2012 to 20.378 individuals at the end of 2015. This shows the start of the decrease after a rapid increase in continental (Near 2 Threatened) white rhino numbers from 1992 to 2010 (averaging +7.1% growth per year) followed by a levelling off coinciding with escalating poaching. (Milliken *et al.,* 2017).

Conservation status

The Southern white rhinoceros is listed as 'Near Threatened' by the IUCN. But the last years the level of poaching exponentially increased which is jeopardising the survival of this species. (Milliken *et al.,* 2017). The Northern white rhino is listed as 'Critically Endangered' as the current population of the sub species is no more than Two individuals, down from an estimated 2.230 individuals in 1960.
By 1977, all African rhino species were listed on CITES Appendix I and all international commercial trade in rhinos and their products became prohibited. However, following a continued increase in numbers, the South African population of Southern white rhino was down-listed in 1994 to CITES Appendix II, but only for trade in live animals to "approved and acceptable destinations" and for the (continued) export of hunting trophies. In 2004, Swaziland's Southern white rhino was also down-listed to CITES Appendix II, but only for life export and for limited export of hunting trophies according to specified annual quota. (IUCN, 2015).

Table ***5. Reported African rhino poaching mortalities 2006-2015***

Country	2006	2007	2008	2009	2010	2011	2012	2013	2014	2015	Total
Botswana	-	-	-	-	-	-	2	2	-	-	4
DR Congo	-	-	2	2	-	-	-	-	-	-	4
Kenya	3	1	6	21	22	27	29	59	35	11	214
Malawi	-	-	-	-	-	-	2	1	2	1	6
Mozambique	-	9	5	15	16	10	16	15	19	13	118
Namibia	-	-	-	2	2	1	1	4	30	90	130
South Africa	36	13	83	122	333	448	668	1004	1215	1175	5097
Swaziland	-	-	-	-	-	2	-	-	1	-	3
Tanzania	-	-	2	-	1	2	2	-	2	2	11
Uganda	-	-	-	-	-	-	-	-	-	-	-
Zambia	-	1	-	-	-	-	-	-	-	-	1
Zimbabwe	21	38	164	39	52	42	31	38	20	50	495
Total	60	62	262	201	426	532	751	1123	1324	1342	6083
Poached/Day	0.16	0.17	0.72	0.55	1.17	1.46	2.05	3.08	3.63	3.68	

(AfRSG, TRAFFIC and CITES Rhino Working Group data in collaboration with range States).

The number of rhinos reported poached in Africa has increased for the sixth year to 1,342 rhinos in 2015, the highest level since poaching began to escalate in 2008 (Table 5). These figures represent minimum numbers as some carcasses may have gone undetected. Poaching in 2015 represents 5.3% for white rhino. These levels are now approaching the average continental growth rates ! (Milliken *et al.,* 2017)

1.2.2 Diet and feeding behaviour

The white rhinoceros is a grazing mega herbivore (Owen-Smith, 1988), consuming large amounts of short grasses and no intake of browse at all (Steuer *et al*., 2010). Their rapid bulk feeding allows them to tolerate food of a lower quality than that required by smaller herbivores (Owen-Smith, 1988). “The natural diet of any rhinoceros species is characterized by a high-fibre and low-to-moderate protein content” (Clauss and Hatt, 2006). Because of the high fibrous diet, they evolved “high crowned cement covered teeth to cope with their feeding demands, as well as a lengthened skull and wide lips” (Owen-Smith, 1973). White rhinos can live up to 4 to 5 days without water (RRC, 2018). Although good quality food is more abundant in the wet season (Pedersen, 2009), white rhinos generally do not over utilize food in their natural habitat (Shrader and Perrin, 2006). The movements throughout the landscapes are influenced by water sources and rainfall (Shrader and Perrin, 2006). In the dry season white rhinos “do not compensate for seasonal declines in food quality by adjusting their food intake rate or diet breadth” (Shrader, 2003; Shrader *et al*., 2006). Shrader *et al*. suggest that white rhinos rely on fat reserves to help them through the period of less quality food. Pederson (2009) states that these animals “are maximising the opportunities to graze on nutritious grasses when they are available”. He concluded that white rhinos are succeeding in exploiting the less than ideal grasslands that they live in. Due to the feeding ecology of the white rhino, grasslands are changed by them into a more suitable habitat for other grazers. (Waldram *et al*., 2008).
Pedersen (2009) found 43 grass species consumed by white rhinos. The nine most consumed species represent 50% of the annual diet. The 20 most consumed species that make up 79% of the annual diet are: *Aristida adscensionis, Bothriochloa insculpta, Brachiaria serrata, Brachiaria xantholeuca, Cenchrus ciliaris, Chloris gayana, Digitaria eriantha, Enneapogon cenchroides, Eragrostis cilianensis, Eragrostis lehmanniana, Eragrostis rigidior, Eragrostis superba, Eragrostis tricophora, Heteropogon contortus, Ischaemum afrum, Panicum coloratum, Panicum maximum, Pogonarthria squarrosa, Schmidtia pappophoroides, Stipagrostis uniplumis.*

1.2.3 Reproduction

Information on oestrus, sexual maturity, copulation, gestation, breeding, birth, delivery and infants is listed in table 6.

Table 6. ***Reproduction facts for the white rhino***

Subject	Details	Reference
Sexual maturity	♂ and ♀ 3 – 6 years	**Goltenboth *et al*., 2001**
Age at birth of last calf	♀ mean: 17.1 years range: 7.2 – 31.1 years ♂ mean: 19.7 years range: 7.2 – 29.2 years	**Fouraker and Wagener, 1996**
Delivery	Labour ± 40 minutes Parturition 10 - 20 minutes Evening or night	**Goltenboth *et al*., 2001**
Breeding season	Peaks in July, Sept and Dec/Jan	**Fouraker and Wagener, 1996**
Birth peaks	April/May, June/July, Nov-Jan	**Fouraker and Wagener, 1996**
Birth intervals	Mean: 30 months	**Fouraker and Wagener, 1996**
	Range: 24 – 48 months	**Grün, 2006**
Oestrus cycle length	27 – 44 days ♂ interested 24 – 48 hours ♀ receptive 12 – 18 hours	**Fouraker and Wagener, 1996**
	28 - 32 (up to 70) days ♂ interested 24 – 48 hours ♀ receptive 12 hours	**Goltenboth *et al*., 2001**
	Short: ± 35 days	**Patton *et al*., 1999**

	Long: ± 66 days	
Copulation	20 - 60 minutes	**Tomasova, 2006**
	30+ minutes Several copulations at peak of oestrus Copulations mostly takes place at dusk or dawn	**Goltenboth *et al.*, 2001**
Gestation period	485 – 518 days	**Fouraker and Wagener, 1996**
	± 490 days	**Grün, 2006**
	515 - 540 days	**Goltenboth *et al.*, 2001**
Age at birth of first calf	♀: 10.7 (5.6–23.5) years ♂: 15.5 (7.2 – 25.2) years	**Fouraker and Wagener, 1996**
	♀ 6-7 years	**Grün, 2006**
	♀ 6.5 - 7 years ♂ 12 years	**Goltenboth *et al.*, 2001**
Calf behaviour	Standing up after ± 15 minutes First nursing 1 - 24 hours after birth	**Goltenboth *et al.*, 2001**
	Begins grazing at 2 months of age, weaning after 1 year and it leafs its mother at 3 years of age.	**Tomasova, 2006**

1.2.4 Behaviour

White rhinoceroses feed and rest alternately during 24 hours. In hot, dry weather they rest during the hottest part of the day. Much of their resting time is spent wallowing to keep cool and to get rid of skin parasites. If no wallowing place is available, they will roll in dust. (Tomasova, 2006) White rhinos may reach speeds of 50 km/h (RRC, 2018).

White rhinos are sedentary, semi-social and territorial. Adult females and sub-adults are rarely solitary. They associate typically in pairs, usually a female with her latest calf. A juvenile stays with his mother for around three years. When the mother calves again, the juvenile seeks another companion, preferably of similar age and the same sex. Stable herds of up to six animals are quite common, while larger groups are the result of temporary aggregations, purpose-made because of availability of favourable food, watering, or resting conditions. Females' home ranges vary between 6-20 km², and usually overlap several males' territories.

The adult bulls are basically solitary and associate only with females in oestrus. Bulls' territories are relatively small, averaging between 1-3 km². The size depends on many factors, including the quality and availability of food and water. Each territory is held by a mature male, often with between 1 - 3 resident satellite bulls. The territory owner ignores these satellite bulls, as long as they behave submissively. Territorial bulls treat foreign intruders far more aggressively than the resident satellite bulls do. (Tomasova, 2006)

At the end of the dry season when water is scarce, some males have to cross other territories on their way to water. This leads to an increase in conflicts and more fighting ensues. Typical fighting wounds seen on male white rhinos, other than obvious lacerations on the head, include broken jaw bones, wounds between the hind legs, punctured abdomens, broken front legs and dislocated hind legs. These wounds are usually fatal. (Pienaar, 1994)

As with the other rhino species, white rhino home ranges are scent-posted with dung heaps placed by both sexes. The collective dung heaps, or 'middens', are usually located at territory boundaries and serve as communication and marking points. All animals add their deposits there, but only territorial males scatter the dung with ritualized kicks and spray urine. (Tomasova, 2006) When a subordinate male gets confronted with a territorial male, it gives a threat display. He lifts its head, roars and makes short rushes at the territorial male. (Pienaar, 1994) White rhinos also communicate vocally, using a wide range of sounds from calf squeaking to snarling or wailing of adults (Tomasova, 2006).

Section 2 - Management in zoos

Recommendations for enclosure designs are always prone to discussion. There is no perfect white rhino enclosure and the below chapter tries to outline as many different options and possibilities, with the welfare of the animal and safety for the keepers as most important arguments. General EAZA standards can be found in Appendix I. These standards are based on present knowledge and practice for the accommodation and care of animals in zoos and aquaria.

2.1 Enclosure

When designing an enclosure, zoo planners should address a lot of general issues to create a safe and easy-to-maintain exhibit (Veasey, 2005). Not only the animals' biological and physiological needs should be taken into account (Curtis, 1982), but the people who daily manage and maintain the exhibits need to be considered as well (Simmons, 2005). The more natural an exhibit looks, the better it is to tell an ecological message. Visitors should not see any bars or mesh, but should have the idea that they are in the habitat of the animal. (Hosey *et al.*, 2009) As stated before, the white rhino is kept in European zoos since the 1950s. They do need a separate in- and outdoor enclosure, as they are not accustomed to the cold winter climate common in most of the EAZA region. Needless to say that an indoor enclosure in a region without these weather conditions could be simplified.

2.1.1 Dimensions

The minimum outside space requirement for 1.3 adult rhinos is at least 1 hectare. But even larger would be preferred. If the space is well structured it could be acceptable to have less space available (e.g. 0.7 ha), but generally this is not recommended. Also, with a smaller enclosure the rhinos can't get away from each other, which can cause problems. Space requirement is also dependant on group composition and space usability /design. With increasing experience (successes and failures) the EEP tries to negotiate the possibilities with all the holders, especially regarding new exhibits. A known fact is that space availability for large mammals in general is important, but surely not the single most important feature.
For every extra animal *0.4* hectare should be added to the minimum space. The outside space must contain separation possibilities as well. It is recommendable to create several enclosures which are connected. This way separate territories could be made for the males and the females and exchange between territories is possible.
The compatibility of the group and the design of the enclosure are equally important as the size. White rhinos are gregarious, but they also like their own space.

The inside enclosure should be at least 30 m^2 per individual (Goltenboth *et al.*, 2001). When a calf is present, an additional 15 m^2 should be available. However, the size depends on a range of factors, like how much time is spent indoors, the proximity to public, etc. More space is always better when possible.
For the inside enclosure it is recommended to have a separate stable for each rhino connected to the group stable. This way the rhinos don't have to go outside when their stables need cleaning.

A surplus facility is needed for animals that can't be placed in the current group. These animals should have their own indoor pen and outside paddock. The surplus - and/or quarantine area should be between 700 and 1100 m^2, depending on group size and length of quarantine. But since a surplus area will be used more often to separate the bull, the recommendation is to make it larger.

2.1.2 Boundary

This paragraph will describe the walls of the pens of the inside enclosures and all primary and secondary boundaries outside. Some examples of boundaries can be seen in figure 4. The advantages and disadvantages of the different boundary types can be found in table 7 (see next page)(Hosey *et al.*, 2009).

***Figure 4*. Examples of primary fencing**

a = wooden posts close together to form a wall, b = horizontal cables with protection, c = diagonal bars, d = wooden posts spaced apart to allow contact, e = horizontal pipes/bars

The bars of an enclosure should be placed vertically or diagonally. At least one side of the pens and corridors must be made out of vertical bars or poles to allow the keeper an emergency exit (Goltenboth *et al*., 2001) and this way climbing can be prevented. Vertical poles are recommended because this way the keeper has several escape routes and isn't blocked by diagonal or horizontal bars.
When using horizontal cables abrasion takes place when the animal is bored, which results in damage of the horn (see figure 5). Also horizontal cables are climbable.

***Figure 5*. Abrasion on the second horn**

Table 7. ***The advantages and disadvantages of commonly used barriers in zoo enclosures***

Type	Advantage			Disadvantage		
	Animal	**Keeper**	**Visitor**	**Animal**	**Keeper**	**Visitor**
Solid *	- Depending on height, provides safety - Prevents disease transmission	- Separates animals - Prevents visitors from feeding animals	-	- Can lead to injury if animals collide with it - Can prevent view of surroundings - May affect communication between animals	- Can prevent view of the animals	- Obstruct view of animals
Partial **	- Can provide greater usable space	- Can aid introductions	- Restricted viewing can make a glimpse of an animal more exciting	-	-	- Can obstruct view, although some materials are less obvious - Considered 'unnatural' - Does not prevent human-animal interaction
Bars	- Can provide greater usable space	- Can facilitate keepers' escape from the enclosure, whether the bars are vertical or diagonal	-	-	-	- Associated with negative connotations of animal welfare - As above
Electric	- Can learn to avoid it	- Easily creates temporary barriers - Cheap	- Good visibility of animal	- Not visible so can get injured - Can get entangled in it - A deterrent, not 'fool-proof'	- Some body parts do not conduct electricity, e.g. horns, hair	- Needs to be well signed as a hazard and out of public reach
Moat	- Rhino may use the water or objects in it	-	- Provides 'naturalistic' view - Invisible barrier between species	- Water can provide a route for disease transmission - A lot of space is required, which cannot be used by the animals in most situations	- Dry moats can flood - Wet moats can freeze - Need method to access animal and/or enclosure safely	- Increases the distance between the visitors and animals, which can reduce visibility

* Materials include: concrete wall, brick wall, wooden posts against each other, etc.
** Materials include: metal posts, wooden posts, cable, etc.

The primary barrier should always have a minimum height of 1.75 m (Goltenboth et al., 2001) and be non-climbable. Any height lower than 1.75 is not recommended.
The barrier can be made out of a dry moat, water moat, posts with horizontal cable or posts without horizontal cables. A barrier of rocks can be used as well and is recommended for the protection of trees and other objects (Goltenboth *et al*., 2001). Preferably don't use rocks as a primary boundary, because there is a chance that the rhinos will climb the rocks.
The primary boundary should only be treated with a non-toxic composition (Goltenboth *et al*., 2001).
The distance between the bars should allow keepers to get through the bars, but the rhino shouldn't get stuck. The ideal spacing is: 20-40 cm between the bars, but this also depends on the type of bars that are used. Standing posts should be 15 cm to 30 cm in diameter and set deep enough into the ground (or in concrete) to avoid individual movement of the poles.

When designing an outdoor exhibit which incorporates a moat (figure 6), the slope must be made out of a non-slippery surface to prevent injuries. On the animals' side the moat should slope gradually, not exceeding 30 degrees, because this part may also be used by the animals (Goltenboth et al., 2001). The wall on the visitors' side of the moat should be at least 1.75 m high (Goltenboth et al., 2001).

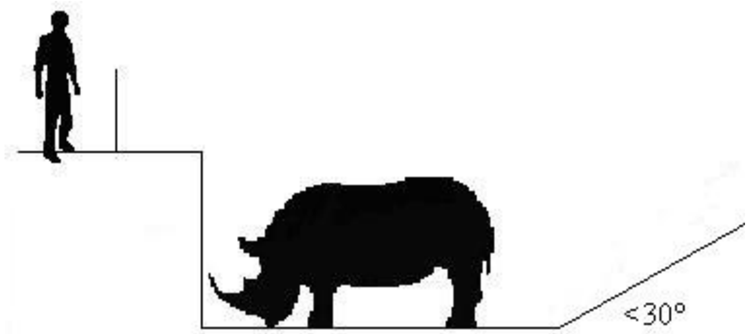

***Figure 6*. Example of a dry moat**

Ditches with strictly vertical walls are considered dangerous and are not suitable for white rhinos, especially with social grouping (Goltenboth *et al*., 2001).
It is recommended to modify existing vertical ditch walls to gradual sloping. As an interim solution, escapes on either side of the ditch must exist (Goltenboth *et al*., 2001).

A secondary barrier can be made out of electric fencing. White rhinos have been seen to retreat from the fence after receiving an electrical shock (Holsey *et al*., 2009). The electric fence should be placed between 0.3 and 1.0 m of the ground. Electric tapes can be used since these are more visible and more resistant. Eventually one could use more than one electric strand.
It is not recommended to use this fence as a primary barrier.

The walls of the inside enclosure should be made out of solid concrete or rock walls. The walls of the inside enclosure should be unpainted since white rhinos will rub against the walls, so a concrete stain is preferable. Waterproofing wall surfaces by covering them with sealant makes cleaning much easier. (Rosenthal & Xanten, 1996)
Each wall of the inside enclosure should be at least 5 m long and the separating walls should be at least 2 m high to give the white rhinos the space they need to move. For separation walls horizontal pipes or cables spaced 25 to 30 cm apart and vertical pipes or posts spaced 25 to 30 cm from each other (Goltenboth *et al*., 2001) can be used. The spaces between the bars should be 20 cm, to prevent a calf from escaping (Goltenboth *et al*., 2001).

One might also consider to construct a separate "calf fence" which can be bolted on the existing fence (see figures 7 and 8).

Figures 7 and 8. Removable calf fence

Separating walls or removable vertical poles can be used to create pens. One pen per animal plus one additional pen is recommended, keeping in mind the size of the desired rhino herd. The possibility to combine these pens to make 1 large area would be ideal. Compatible females can share a pen, but holding pens should be available to be used if necessary. Males cannot automatically be housed together with the females in the same indoor facility. The pens should be large enough to accommodate more than one rhino.
It is recommended that at least one opening can be moved per pen combination. Solid door panels can be used as a visual barrier and bars can be used to allow visual contact (Goltenboth *et al*., 2001).

The inside enclosure should always have more than one entrance to prevent animals from getting trapped by others. An escape route should always be available.

Doors should be reinforced with strong hinges and locks (see figure 9). They should be constructed of either heavy metal, galvanized steel, pipe that is either hinged or sliding, or of wood reinforced with steel. The bottom part of a door should be reinforced with steel plates to minimize possible damage. (Goltenboth *et al*., 2001)

Figure 9. Strong steel door hinges

Figure 10. Crush or restraining area

Connecting pathways and tunnels between indoor/outdoor facilities should be approximately 1.5 m wide for an adult animal to pass freely without feeling trapped, but to make it more difficult for the animal to turn back and get stuck. Long pathways should have bar doors to be closed after the animal has passed through. There has to be a passage next to the rhinos for the keepers, so they don't have to walk behind the rhinos. This way one can also make simple crushes or

restraining areas (see figure 10) where the keepers have safe and easy access to the animals for husbandry purposes.
Outdoor facilities must have at least 1 entrance/exit for heavy trucks or cranes and the indoor facility preferably as well. (Goltenboth *et al.*, 2001)

For the keepers it is important to have access to the enclosure all the way round. This way they can easily perform medical examinations and monitor calves.
For the gates it should be taken into consideration that the keepers don't get into contact with the rhinos. This can be done by installing sliding doors instead of revolving doors.

2.1.3 Drive-through enclosure

White rhinos are often kept in drive-through enclosures, since a drive-through has a high educational value for the visitors and the species is usually negligent towards visitor cars.
It is possible to keep white rhinos in a drive-through enclosure, though there is always a risk with rhino charging visitor vehicles.
The character of the individual rhinos, whether the animals are accustomed to vehicles, the size of the enclosure, the type of vehicles, etc. are all factors influencing the suitability of an individual white rhino for a drive-through enclosure. Continuous surveillance by keepers is recommended in these drive through exhibits to keep control over animal and visitor.
The gates in a drive-through enclosure can consist out of a sluice, cattle grid or electric floor mat and should be manned by a gate marshal or keeper with possibilities to intervene in case the rhino poses a threat towards the cars of the visitors. A contingency manual gate should be constructed as well.

2.1.4 Substrate

White rhinos can suffer from nail cracks and laminitis (see chapter 2.7.1). This means that the substrate they are walking on is very important. The best types of substrate for the stables can be a combination of wood chips, concrete, rubber, soil and sand (see figures 11 and12). One could choose to divide the stable up into a hard and a soft part (see figure 13). Especially stables designed for females to give birth are

***Figure 11 and12*. Complete "biofloor" stable**

***Figure 13*. Combination of "hard" and "biofloor" stable**

recommended to have a soft part, for instance a sand-box, as this is much more comfortable and less slippery for the calf.
Hay can be put on top of the concrete to offer a softer place to lay down (IRKA, 2010a).

Attention needs to be given to good drainage when using "biofloors". Either by having drainage systems under the "deep litter biofloor", or natural draining soil. If for example the natural soil is sandy, one could add a metre of small pebbles as "draining bed" and on top of this the deep litter wood pine. Keeping "biofloor" humid will increase the functionality and longevity of this type of substrate.

Floor heating can be installed in a minor degree, but caution should be taken, especially because recent experiments with substrate in stables are very promising considering animal welfare. The combination of soft substrate stables with floor heating off course is very problematic. If one chooses part of the stable to be used for soft substrate one might choose for floor heating in the rest of the stable. An advantage of floor heating is that the stable dries up nicely which is beneficial for the hooves of the animals, but if floor heating is used too extremely it may cause damage to the hooves. Heating wise there is not really a necessity for floor heating.

For the outside enclosure a self-draining surface that provides adequate footing is recommended (Goltenboth *et al*., 2001). The substrate in the outside enclosure should be made out of a combination of grass, sand and concrete. The concrete part can be used to place food on and this makes it easy to clean for the keepers.

When the quarantine area is being used to separate animals, the substrate should be the same as in the normal enclosures. When the area is only used for quarantine, the substrate should consist out of materials which are easy to clean, such as sand and concrete.

2.1.5 Furnishings and maintenance

Furnishings

Furnishings are installed in the enclosure to increase the natural behaviour and reduce stress levels. Also for management purposes additional furnishing can be installed, like a scale or separation walls.

White rhinos need a pond and/or mud wallow (see figure 14) for skin health, temperature regulation and behavioural enrichment. White rhinos are less likely to use ponds since they don't swim, so pools are not necessary and can sometimes be a hazard if they are too deep (IRKA, 2010c, Goltenboth et al., 2001). Mid deep mud banks and natural ponds should be sufficient. The white rhinos will construct their own mud wallow when given a start. A mud wallow needs to be renovated once a year (Goltenboth et al., 2001) or at own insight, to prevent the mud wallow from getting too big and too deep. When a pond is constructed it should have a depth of max 0.5 to 0.7 m.

Access to shade and protection from rain is a must. By giving shaded areas to the white rhinos they can rest in cooler areas during the hotter periods of the day or when the stable is inaccessible (Goltenboth *et al*., 2001). Possible options are trees and/or other vegetation and roofs as artificial means. It is recommended that a number of adequate sun and rain protection zones are provided. Sun shelters can also be used as rain shelters, trees do rarely serve this purpose. Wind protection should be provided in some parts of the enclosure, unless a solid wall barrier already exists (Goltenboth *et al*., 2001).

***Figure 14**. Two white rhinos "bathing" in a Mud wallow*

Another consideration may be scratching posts or poles. One might consider using these for visual attraction as well and include them as barrier since these look far more natural than steel bars (see figure 15 and 16).

***Figure 15 and 16**. Scratching poles as "barrier"*

It can be important to have visual barriers present in the enclosure (depending on the size of the enclosure), so the rhinos can escape from each other's eye sight. Also visual barriers between rhinos and other animals can be beneficial, especially during an introduction.

Maintenance

Easy maintenance of the enclosures is worth considering when building enclosures. Well-constructed enclosures are easier to maintain and are subsequently more likely to be responsibly managed (Rosenthal and Xanten, 1996).

The indoor housing should be cleaned every day. This can be done with a high pressure hose/fire hose, sometimes using disinfectant or soap, depending on what type of stable/substrate. It should get mucked out daily, but to fully clean it with water on a daily basis is not necessary.
The outside enclosure and enrichment objects should be cleaned when needed.
To make things easier while cleaning, it is recommended to leave a small pile of dung on a desired spot after cleaning. This way the rhinos will use that place as their dung site.

2.1.6 Environment

Temperature

White rhinos in captivity live in a variety of climates. A heated stable with enough room for exercise is needed in cooler climates (IRKA, 2010a). White rhinos should have access to their inside enclosure when the temperature is <5°C, depending on the weather conditions. When there is rain, snow, hard wind or hail at a temperature of respectively <10°C, the rhinos should have access to their inside enclosure. Snow and cold are not harmful for healthy rhinos, but they are less tolerant of wet and cold/windy conditions. A minimum outdoor temperature of around 12 ºC is required to let the rhinos stay outside all day and night (Goltenboth *et al*., 2001), depending on the weather conditions.

The temperature in the stable should be at least 14°C with the capability of maintaining some areas at 20 °C as maximum (Goltenboth et al., 2001). For sick or older animals the inside temperature should be a little higher than for healthy white rhinos and possibly reinforced with a heating lamp.

Heating should be available, of which radiant heating is recommended over floor heating (see 2.1.4). With the changing initiatives for soft substrate stables, floor heating should be used less and less. But in combination should be restricted to cover no more than one quarter of each pen (Goltenboth *et al*., 2001) as otherwise it may cause problems by raising dust and ammonium fumes The temperature of the floor heating should be between 10 and 14 ºC.

Humidity

The humidity in the stable should be kept between 40 – 70% .

Ventilation

Adequate ventilation must be available. Natural ventilation together with fans and air exchangers is recommended, including exhausters in the roof, in the walls or near the floor, for effective removal of ammonium fumes. Draught should be avoided. (Goltenboth et al., 2001)

Lighting

It is unsure whether the period of (day)light should be kept at 12h, also during the winter months. Especially because white rhinos experience great seasonal differences in light in their wild habitat as well. The lighting should consist of natural lighting combined with artificial lighting. The use of UV lamps can be beneficial for the animals, but this depends on the type of lamp and the distance to the animal. It can be important for young animals with regard to bone structure, but it is practically very difficult to use since the distance between the UV light and the animal should not be too great. During the winter UV-therapy for 1 hour can help to prevent skin problems. The lux index should be between 950-1000 lx.

Quarantine

When designing a quarantine area, the following aspects should be kept in mind; disinfection of the floors should raise no problems, the floor should not be slippery, floors of well-draining tiles that also hold back water are recommended and rubber matting for hoofstock may be used (Goltenboth et al., 2001). It is also important to have an easy method for waste removal , extra heating for sick animals, independent water and drainage, possibilities to take samples without sedation and ease of cleaning. Finally the quarantine box should be separate from the rest of the stables and the animals.

2.2 Feeding

A diet should always be balanced and contain the required energy and nutrients. The age, sex and general health of every individual animal should be considered when designing a nutritional program. Therefore the basic diet, special dietary requirements and non-nutritional aspects of feeding, as well as methods of feeding and information concerning water supply are described. In §1.2.2 diet and feeding behaviour of the wild white rhino can be found. “The software programme Zootrition or any similar spreadsheet calculations should be consulted where possible to analyse nutritional quality and quantity of food consumed and wasted” (EAZA husbandry guidelines format, 2008).

2.2.1 Basic diet

As stated in Section 1, the white rhinoceros is a grazing mega-herbivore (Owen-Smith, 1988), which means that they eat large amounts of short grasses and no browse at all (Steuer *et al*., 2010). It is rarely possible to provide natural grass species for rhinos in captivity and therefore a good understanding of the nutritional requirements of white rhinoceros is necessary when formulating captive diets. In the wild, white rhinos have a diet that contains a high fibre content and a low to moderate protein content (Clauss and Hatt, 2006). On average a white rhino should spend at least 50% of the day (Owen-Smith, 1988) on foraging/feeding (see figure 17), but this strongly depends on the time of year.

***Figure 17*. White rhinos grazing in Greater St. Lucia Wetland Park, South Africa**

Nutritional content

Due to similarities in digestive tract morphology, the domestic horse (figure 18) represents the best nutritional model for all rhinoceros species (Stevens and Hume, 1995). Diets should be formulated using current recommendations for horses of various physiological stages. There are however differences reported in the requirements of fat soluble vitamins.

According to the diet sheets of zoos, the food items listed in table 8 can be given to white rhinos. However, Clauss and Hatt (2006) stated that there is no nutritional or financial rationale for offering fruits or vegetables to white rhinos. “If a fruit component of the natural diet is to be mimicked for pedagogic or emotional reasons, then commercially available green leafy vegetables best resemble ‘wild fruits’ in their nutritional composition. Onion, brassica and rape should be avoided, as they have all been linked with haemolytic anaemia in other species.” (Clauss and Hatt, 2006) “An adult white rhinoceros of 1800 kg can cope with a daily intake of 25-35 kg of dry matter. The high moisture content of fresh foods would mean that four times this weight would probably be consumed in a 24-hour period.” (Jones, 1976) “Commercial fruits, vegetables, cereals and grain products should not be fed, except for medication or training

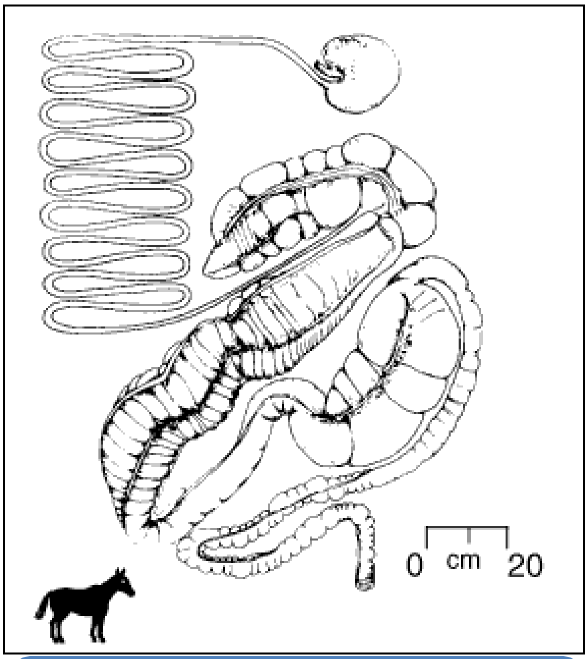

***Figure 18*. Domestic horse (Equus caballus) digestive tract**

purposes. Although even in these cases, green leafy vegetables are to be preferred" (Clauss and Hatt, 2006).

Table 8. ***Possibilities for food items and quantities per adult white rhino per day***

Food item	Quantities per adult rhino (kg)
Hay	Ad libitum – 2 to 3 bales (50 to 75 kg)
Lucerne	–
Alfalfa hay	30-40 kg
Grass	Ad libitum
Rye	–
Clover	–
Avena	–
Fruit	0 - 2
Apples	20 kg – 1 to 2
Vegetables	2
Carrots	1 to 2
Beetroot	–
Beta vulgaris	–
Mixed chopped carrot / celery / turnip / kohlrabi	20
Pellets	1 – 6
Dry bread	–
Flaked maize	–

Quantity, quality and frequency

Because of the large amounts of grass eaten by white rhinos, lower quality grass is tolerated, in comparison with smaller herbivores (Owen-Smith, 1988). "Maintenance requirements of hindgut fermentators should be 0.6 MJ digestible energy per 0.75 kg metabolic body mass" (Clauss and Hatt, 2006). Dierenfeld (1999) stated that dry-matter intake of an adult rhinoceros ranges from 1 to 2.5% of body mass in zoo studies. Approximately 1.5% of body mass (on a dry matter basis) is advised, but this varies per season. It should be noted that if you give 1.5% of food (dry matter basis) this means you should give nearly all of that amount as forage, not fruits or bread. When the condition of the rhino drops, you shouldn't increase the energy density of feeds (like using bread or grains), but increase the amount of feeds with roughage.

Food should be offered two or three times a day with constant access to grazing (depending on seasons). Hay must be available ad libitum and stored at the same temperature as surroundings prior to feeding. "Rhinoceros are prone to obesity and food should be given restrictively, based on either the results of regular weighing or regular assessment of the body-condition score. Changes to the amount of roughage offered should be accompanied by corresponding changes to the amount of pelleted compound feeds offered" (Clauss and Hatt, 2006).

Roughage

Hay of appropriate quality should be the major part (in quantity and nutrients) of a white rhinoceros diet (Clauss and Hatt, 2006). The quality of hay needed depends on the availability of good grazing and the amount of concentrate fed. Care should be taken with very high quality legume or small grain hay, as it is very easy digestible. Hay quality should be monitored by laboratory analyses and judged by sight to ensure appropriate quality and to determine what additional nutrients are needed for a balanced diet. According to Clauss and Hatt (2006) grass hay is the appropriate roughage for white rhinos, but to ensure adequate protein levels, 20% of the hay offered should be legume hay. In §1.2.2 the twenty most consumed grass species in the wild are listed. Suitable plant species for feeding are most non-evergreen tree species like willow, silver birch, sycamore, grass and hay.

Pellets

Concentrate feeds should only be used to balance energy, protein, minerals or vitamin needs. “It should only be used to satisfy energy needs when adequate roughage is not available. There is no scientific rationale for the inclusion of grain products in pelleted compound feeds for strict herbivores” (Clauss and Hatt, 2006). Alfalfa pellets are not recommended (Goltenboth *et al.,* 2001). According to Dierenfeld (1999), not more than one-third of the overall calories should come from pellets. Large horse feeds or high-fibre ungulate pellets (>1.0 cm diameter) work well with white rhinos. Pellets should be given in at least two feedings daily for better utilization and, when practical, a small feeding of hay should be encouraged prior to each concentrate feeding.

Supplements

Dietary supplements are unnecessary in properly formulated rations. Daily vitamin E supplement is necessary if fresh grass or grazing pastures are not available. Salt blocks can be used.

Calcium (Ca) and phosphorus (P)

“A diet based on any hay (grass or lucerne), supplemented with pellets, does not require any additional calcium source” (Clauss and Hatt, 2006). Different calcium levels in forage in the natural diets of white rhinos are shown in table 9.
“Roughage based diets are particularly vulnerable to phosphorous deficiency. Hypophosphataemia (low levels of phosphorus in the blood) has been observed in rhinoceros with haemolytic crises, so a deficiency of this mineral in the diet should be avoided.” (Clauss and Hatt, 2006)

Copper (Cu) and Zinc (Zn)

Clauss and Hatt (2006) stated that some diets for rhinoceros are deficient in copper. Further research into copper metabolism in white rhinos is needed. Zinc deficiency may lead to the development of skin and foot lesions, so zinc should be supplied according to the domestic horse recommendations (table 9) (Clauss and Hatt, 2006).

Table 9. ***Mineral requirements (in g/kg DM) of free-ranging rhinos compared to domestic horses***

Mineral	Temperate		Recommendations maintenance level	
	Lucerne	Grass	Rhino	Horses
Ca	21.0	4.8	2.4	2.4
Cu	0.011	0.006	0.004	0.01
Fe	0.180	0.129	0.177	0.04-0.07
K	22.0	21.6	8.5	3.0-6.0
Mg	2.8	1.5	0.8	0.9
Mn	0.040	0.074		0.04
Na	1.1	0.05	0.3	1.0
P	3.0	2.7	1.0	1.7
Zn	0.024	0.019	0.023	0.04

2.2.2 Special dietary requirements

As said before, a diet should always be balanced and contain the required energy and nutrients. The age, sex, reproductive status and general health of every individual animal should be considered when designing a nutritional program.
During pregnancy and lactation the dam should get the same food items and amount as normal, but the forage should be available ad libitum.
One could consider the increase of protein, for example by giving about 20% of the roughage as lucerne hay.

After weaning, the dam gets the same food as normal. This can be done by gradually reducing the diet amounts back to normal. If the dam has been fed roughage ad libitum all the time, you should observe her to check whether she is gaining weight once weaning takes place. The weaning can happen forced or naturally. If the condition of the dam (you might want to let her have ad libitum roughage to replenish) is back to normal, the roughage can be reduced. But this is probably not realistic, as she will have a growing calf next to her.
Hand rearing of rhinos is executed in accordance with the known procedures for other species. Hygiene is an absolute must to avoid contamination of the milk, as well as intensive care by one or more keepers. The applied milk preparation should simulate the natural mother milk. Skimmed, pasteurized or homogenized milk (3.2% fat, 3.3% protein and 4.7% lactose) is well suited. Horse milk has a very similar composition, so this is also suitable for hand rearing.
Supplementation of vitamins and minerals is recommendable. If possible, colostrum should be given within 24 hours after birth, or alternatively rhinoceros serum. For hygienic reasons and to avoid hasty drinking, the young should rather be bottle fed than with a bucket." (Goltenboth *et al.,* 2001) "The quantity fed should range from 10 to 13% of body weight. Animals should be fed every 2 hrs. Because infants suckle during daylight hours, feeding should be equally spaced in a 12-hr period, not to exceed 3% of body weight at any one feeding. It is recommended that feeding begins with 10% of the body weight, split equally into 12 feeds of 1 or 2 hours apart, during daylight hours. The quantity of formula fed should be adjusted daily based on the animal's weight. Animals should be weighed at the same time each day. Fresh water should be available at all times." (Fouraker and Wagener, 1996). "Solid food should be available from birth on, as calves will nibble on solid food very quickly. A nutritionally complete pelleted diet such as horse feeds or high fibre ungulate pellets, in addition to alfalfa hay, is appropriate. Formula may be decreased by gradually eliminating the number of feeds or decreasing the amount offered per feed" (Fouraker and Wagener, 1996).

2.2.3 Method of feeding

To avoid ingestion of sand, which can cause colic, white rhinoceros should not be fed on sandy grounds (Clauss and Hatt, 2006). The food can best be placed on the floor, on a straw bed, in racks low to the ground or on concrete. Schmidt and Sachser (1996) researched food dispersal and behaviour in white rhinos. They found more agonistic encounters when hay is provided in one pile for all animals, in comparison to one separate pile for each animal. "Stress-hormone levels were elevated during clumped feeding and agonistic behaviour continued to be observed long after the hay was consumed." These results underline the importance of providing an appropriate number of feeding places for animals that are maintained in social groups. Depending on size of feeding stations, the number of feeding stations outdoors should be at least the same as the number of individuals. These feeding stations should be far away from each other to minimize stress and competition.

2.2.4 Water requirements

A fresh water source at room temperature should be accessible at all times (Goltenboth *et al.,* 2001). Troughs or automatic drinking troughs are the best ways to provide drinking water indoors to white rhinos. Outdoors troughs, automatic drinking troughs or a moat or pond can be used. If a moat or pond is used, its hygienic status must be checked regularly. Self-operating water sources are not recommended (Goltenboth *et al.,* 2001). Make sure the horn does not obstruct the drinking behaviour of the rhino.
The troughs must not be too high, too narrow or too deep. It is also not advised to place them against a solid wall, because this can obstruct their drinking. The best place to put the water trough is in the middle of an enclosure, so the rhinos can't use the trough as a step and they aren't obstructed in their drinking behaviour.

2.3 Social structure

In the chapter social structure, topics such as group composition and introductions are discussed. When animals are introduced into an existing group, the introduction can be stressful for animals and keepers. To keep stress levels as low as possible certain steps can be taken.

2.3.1 Basic social structure

An optimal sex and age structure for a new group would be 2.3 adult animals with their calves (Pienaar, 1994). Some evidence from captive white rhinos in zoos indicates that cows can come into oestrus more difficult if there is only one bull (Lindeman, 1982), although this is not always the case in free-ranging populations. It is always a good policy to have at least two bulls in a population just in case one gets injured (Pienaar, 1994) There always should be a separate inside and outside enclosure for the remaining bull. A downside to having two bulls is the problem that one of the bulls has to stay in the stables the whole time if there is no second exhibit, so it isn't always a good solution.
The optimal group size depends on many different factors, like exhibit size, separation possibilities, temperament of the individuals, social experience etc. White rhinos are social animals and for that reason multiple female groups are recommended. To house multiple males may have benefits, but generally if you have one exhibit this will not be ideal. Theoretically it would be the best to provide both males with a separate territory.
Males can be housed separate or visually isolated, but appear to prefer to be housed with at least one female. Females should be housed in the best social setting as possible.
At an age of 2-4 years white rhinos are ready to be taken out of their old group and can be introduced into a new group. Until a calf is about 3 years old it should be kept with its mother. If an animal is introduced too young, it is possible that it will look at the other rhino's as siblings and breeding is less likely to occur. There is also a chance of social suppression; the young animal can be socially suppressed by the older females and become a flat liner.
The best moment to take a calf away from its mom is the time when the mother starts to back away from her calf. At this moment in nature the calf starts looking for its own herd. The best way to determine this moment is by observing their behaviour.
Some calves are mature at a very young age and some mature much later. This varies per animal and per situation.

2.3.2 Changing group structures

A flexible rotation system where the group composition differs each day gives the white rhinos stimulation. When extending the group structure it is necessary to provide ample space and hide-outs.

The most important elements in introducing individuals for breeding are the age and social status, which should harmonize. For instance, a sub-adult male will not know how to approach an adult female, subsequently the female will fight or intimidate the male. A proven breeder male under normal circumstances will breed with a young female. However, it should be noted that successful introduction of individuals is largely dependent on the introduced animal's personality. Either sex of captive individuals have shown aggression during introduction. (Goltenboth *et al*., 2001) For 'training' a young male may be introduced to a young female, whereas an older male will be brought into action to fertilize her. (Goltenboth *et al*., 2001)
When wanting to introduce an animal it is important to know its background. For instance, it would be more difficult to get a rhino that has always lived in a Safari park adjusted to a zoo, than the other way around.

Introduction

When an animal needs to be introduced to a group, it is important that the introduction is done gradually.

During the introduction of an animal, the following steps may be taken:

- Provide auditory, olfactory, and visual contact between the newcomer and the herd.
- Provide tactile contact through bars with a few members of the group.
- The individual is first introduced to either the dominant animal or the entire group.
- During the introduction a veterinarian as well as experienced staff members should be present with a high pressure water hose or a car, to intervene (Goltenboth *et al*., 2001).

All animals should independently get to know the enclosure and each other through bars. It is recommended to first group the females and to introduce the male later. During an introduction all animals will show the same known behaviour patterns like aggression, greeting, fighting and bluff charging. The newly introduced animal may show fear and develop diarrhoea. (Goltenboth *et al*., 2001)

During the introduction the following signs are good indicators that the new animal is ready to be introduced to the rest of the group: reduced aggression, flehming, feeding normally and lying down relaxed, friendly, curious behaviour. Social suppression should be kept in mind when introducing new animals. Integration is reported to take between 1 to 10 weeks, but this isn't an actual guideline. It depends on the animals how successful the introduction is and some rhinos never really adjust to the new situation. (Goltenboth *et al*., 2001)

Excessive aggression is a signal that the introduction is not going according to plan and that it can go wrong. When this happens it is best to separate the new animal (not total isolation) and maybe try to reintroduce it again later. (Goltenboth *et al*., 2001) A distraction during the introduction is a good idea to calm the animals down, for instance food.

Introducing a female in a female group

Prior to first contact, the female should be familiarized with the outdoor enclosure. Indoors, females should have eye contact through bars. One could choose to get the new rhino introduced to the "calmest" rhino from the group and continue from there. Before the final introduction they should no longer show aggression. Introduction while a female is in oestrus can be beneficial. Close observations should be continued after the introduction. (Goltenboth *et al*., 2001)

Introducing a male to a female group

Male rhinos are territorial, they mark their territory with faeces and urine, scraping hind legs and spreading dung. A male's marking should not be removed prior to introduction of the female group. Females tend to form strong pair bonds, even if not related. In some cases this has interfered with breeding, when the female's cage mates keep driving off the approaching male.

A second male kept in an adjacent enclosure may stimulate breeding through potential competition. (Goltenboth *et al*., 2001) The second male can be held in the surplus area and when necessary be introduced to the group of females for mating. A possible disadvantage of such a system can be that there is a risk that the second male is more busy with establishing his territory than with breeding. Another disadvantage is the fact that you don't know which male is the dominant one before introducing them. This can also lead to social suppression.

Upon introduction of a male to a group of females, the females should be together and comfortable. The new male will exhibit aggression, greeting, bluff charging and fighting. The females in return will show the same. (Goltenboth *et al*., 2001)

It is not a good idea to do a male-female introduction while a female is in oestrus, because of the continued interest of the male in the female. Integration is reported to take 5 weeks (Goltenboth *et al*., 2001), but a full, stable integration might take much longer. Male rhinos are often tolerated but not really accepted.

2.3.3 Sharing enclosure with other species

There are advantages to keeping white rhinos in a mixed species enclosure, such as education and behavioural enrichment. The educational value is that the visitors can see the rhinos living with other species in the same habitat. The enrichment factor for the animals is that they can be distracted by the other species.
The keywords for mixing species is a lot of space and enough fleeing possibilities.

Sharing species

The following species are known to be kept with white rhinos (see figure 19): mongoose, monkeys, (plains) zebras, giraffe, crowned crane, ostrich, duck, geese, eland, lechwe, camel, cheetah, ostrich, waterbuck , wildebeest, greater kudu, springbok, blesbok, watussi, nyala , (congo) buffalo and gemsbok. When animals are breeding in the enclosure there should be enough space and hide-outs (Goltenboth *et al*., 2001).

Separated species

Several species have been described as possible problem species for a mixed rhino exhibit. These are: elephants, addax, zebra stallions, wildebeest bulls and giraffe bulls. Mixing those species with white rhinos can be very risky even in large enclosures.

***Figure 19*. Rhino, ostrich and zebra in mixed exhibit**

2.4 Breeding

Breeding is an important component of conservation. The captive population serves as a potential reservoir for reintroducing animals into the wild. However, reproduction in white rhinoceroses, especially those in captivity, has been distinctly disappointing. Reasons for this are still unclear. The understanding of the reproductive biology of the white rhinoceros is still limited and details are largely unknown." (Goot, 2009) "From the first descriptions of the reproductive anatomy and oestrous cycle to the present use of advanced assisted reproduction technologies, researchers and veterinarians have attempted to understand the function and dysfunction of the reproductive biology in this species (Hermes and Hildebrand, 2011). Many of the founding population, given appropriate husbandry and management, reproduced well, but reproduction among captive-born females has been extremely sluggish" (Swaisgood, 2007). This chapter outlines breeding, including details on mating, pregnancy, birth, development and care of young and procedures involving hand-rearing. The contraception policy is also described. See §1.2.3 for more reproduction information.

2.4.1 Mating

White rhinos are seasonal or opportunistic breeders (Skinner *et al.*, 2006). This results in the possibility that a calf can be born all year round. Both genders need to be sexually aroused. By rotating females to different bulls and the bulls not having constant access to the cows, a sexual tension field can be created. When a cow approaches oestrus there may be noticeable signs like male interest, squirting urine, vulva wink, aggression, restlessness, inappetence and standing still. Faecal analysis on hormone levels can be done in Vienna (Dept. of Natural Sciences - Biochemistry University of Veterinary Medicine). The following behaviours are commonly seen during courtship: male constantly following female around, approaching, spray urinating, smelling at urine and faeces, over-marking, male smelling females' back end, dragging, flehmen, vocalisation, body and horn rubbing and pushing, ambivalent behaviour, erection, mounting, chin resting, heavy breathing, female standing still, female backing up to the male and frequent urinating and wetting herself. With ambivalent behaviour the danger of separating too early is present. Copulation lasts for 30 minutes or more and will occur several times at the peaks of oestrus. A male remains mounted for 19 minutes or much more" (Goltenboth *et al.,* 2001).

2.4.2 Pregnancy

White rhino females display two different types of oestrous cycles. One which lasts for about one month and another lasting for about 10 weeks. The two oestrous cycles (Schwarzenberger et al, 1998; Patton *et al*, 1999) can occur within the same female. The shorter cycles are regarded as potentially fertile and are most common.

***Figure 20**. IZW scientist Dr. Robert Hermes confirming pregnancy by carrying out a sonography*

A pregnant white rhino will gain weight and undergo physical changes like a swollen vulva, udder growth, and a genital oedema (swollen outer genital) shortly before parturition. She can also undergo behavioural changes, for instance less tolerant towards herd mates, avoiding the male and being aggressive towards him. Those changes mostly aren't visible until the end of the pregnancy.

Pregnancy should be confirmed by the measurement of an elevated progesterone concentration in the faeces or blood. Progesterone indicates pregnancy reliably 3-6 months after mating. Ultrasound (see figure 20) can detect pregnancy from 2 to 4 weeks after conception (Hermes and Hildebrand, 2011) and might be used when hormone results are inconclusive. When a female is pregnant, one could think of special husbandry adjustments listed in table 10. Those adjustments should happen gradually to prevent high levels of stress during the pregnancy.

Table 10. ***Possible special husbandry adjustments during pregnancy***

Husbandry adjustments
Not overfeeding the female.
Separate female during the evening from others if becoming stressed or less tolerant.
Close down gaps in vertical bars.
Deep litter calving pen.
All round keeper access if possible and reduce water level in trough.
Possible extreme aggression from the bull during the pregnancy should be monitored.
The female should be used to stand separately prior to birth, in a stable with bars in between her and the group. It is important that she is not isolated from the group but does have more relaxation on her own.

Artificial insemination
When natural reproduction cannot occur or doesn't result in viable offspring, artificial insemination (AI) can be a solution. The first successful AI in a rhinoceros was reported in 2007 using fresh semen. The first successful artificial insemination with frozen-thawed semen in rhinoceros holds great promise for the future of the mega herbivores. Semen samples can be collected and preserved from both wild and captive populations (Hermes *et al*, 2008). Artificial insemination is an option when breeding is desirable and natural breeding is not successful and all other possibilities have failed (Leibniz Institute for Zoo and Wildlife Research, Berlin, Germany).

2.4.3 Contraception

Managing a rhino so that it is unable to breed is the only form of contraception at this point in time (e.g. separation and/or transport). However, there are old individuals which might require contraception for clinical reasons. Sometimes it is better to make a female infertile for animal welfare reasons, for instance extensive tumour growths, regular blood loss, chronic endometritis and necrolysis of extensive tumours. Such geriatric treatment/contraception program is currently coordinated by the IZW (Leibniz Institute for Zoo and Wildlife Research, Berlin, Germany).
The choice has been made that birth control, in the form of surgery, hormone suppression, etc., is not an option for this species because of the low population numbers and birth rate in European institutions. Breeding is desirable to get a viable and stable ex situ population. Managing the individual white rhinos in a way that they can be used optimal for the breeding program is of vital importance. (Versteege, 2010b). More important, these birth control measures are too invasive to ever be considered practical.

2.4.4 Birth

When a white rhino female is at the beginning of parturition, the following behaviours and characteristics can be seen: she will retreat to a quiet place, refuse to eat, urinate frequently, pace, getting up and down and generally looking uncomfortable, swelling of the udder and vagina, discarding of mucus, defecating, twisting of the tail in an upheld position and hard drops of excretion from the udder. Many females show different behaviours. Dropping of progesterone level is a good sign that within 36 hours the calf will be born. When these signs are seen the female should be separated from the group, but not completely isolated, with bars in between the female and the rest of the herd. When the female is separated, minimal olfactory, auditory and visual and no physical

contact with the male is recommended. However, olfactory, auditory, visual and physical contact (through bars) with other females is recommended.
Whether the birth should be observed by a person or camera to monitor the process is an institutional decision.

Delivery (see figure 21) is of short duration; labour lasts about 40 minutes, parturition time about 10 to 20 minutes (either standing up or lying down). Normally delivery occurs in the evening or at night. For the infant to be able to stand up it is necessary to prepare a none-slippery floor surface, for instance a sand box. The placenta will be discharged right after giving birth and in many cases will be ingested by the mother. Suckling is done either standing up or lying on the side. The latter may result in the infant falling asleep. First nursing is seen within less than 1 hour or within 24 hours after birth. First standing up by the infant is commonly seen after 15 minutes. It may however take 1 to 2 hours. (Goltenboth *et al.,* 2001)
Whenever possible or desired, neonatal examinations can be performed. These can include a weight check, a dipstick blood glucose, total solids, CBC, sera chemistry profile, sera/plasma for vitamin-E levels and, when possible, stored sera. Examinations may include vitamin supplementation and the placement of an identification transponder (Fouraker and Wagener, 1996). In general within the EEP it is agreed that neonatal exams should only be performed in case of emergency. Only some exams, like a visual check and weight checks should be performed in the first weeks. The identification transponder can also be placed at this time.

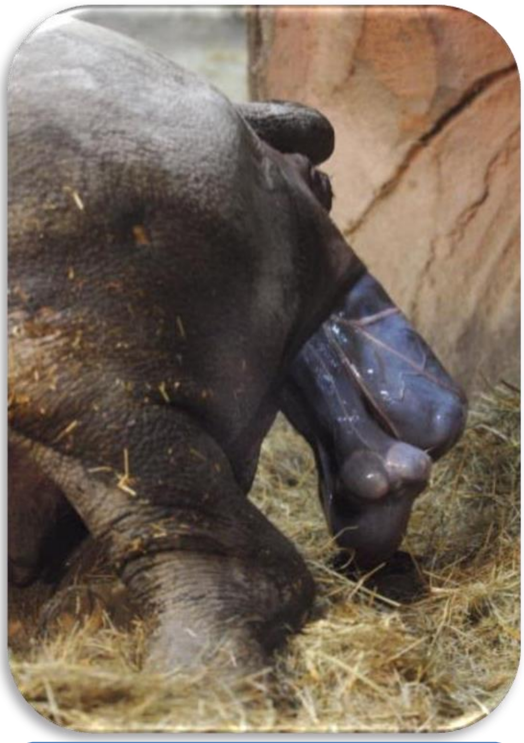

***Figure 21**. A southern white rhinoceros gives birth to her calf conceived from AI in Budapest*

2.4.5 Development and care of young

To prevent the dam from killing the calf it is recommended to create a calm environment, routine, close observation, all round keeper access to the pen and enrichment. When everything goes as planned the calf can be introduced to the male/herd in 2 weeks after birth. This depends on the temperament of the female, the group and its relationship with the male and also on the temperature (harsh European winters). If the mother neglects her calf, the best solution to help the calf is to foster and hand-rear it. When hand rearing is necessary, it's best to do this near the mother or other rhinos. Young rhinos can learn to suckle out of a hanging bottle rather quickly and this way they can be kept in with the mother if she remains calm. It should be avoided to completely separate the calf and hand rear it without the presence of other rhinos.

***Figure 22**. Keeper bottle-feeds white rhino calf*

2.4.6 Hand-rearing

Hand-rearing (see figure 22) becomes necessary when the young is completely rejected by the mother, when medical problems exist in the mother or the infant, or when the infant fails to nurse. Hand-rearing an infant must be considered very carefully. Should the female show aggression towards the young or have medical problems, hand rearing can be considered. (Goltenboth *et al.,* 2001) See §2.2.2 for feeding instructions. Hand rearing should only be seen as an emergency solution.

2.4.7 Population management

For a healthy ex situ population the target population for the white rhino is set on 350 animals by the EEP (Versteege, 2010b). Analyses of the 2006-2007 EEP population indicate that more than 30% of the population is more than 35 years old, but that only 26% of the white rhinos imported since the 1950s ever bred, and only 13% of the F1 generation has bred to date. More recently, a much better understanding of the natural history of these animals has resulted in improved management. Zoos holding white rhinos within the EEP are now strongly encouraged to hold larger groups. The realization that reproduction of daughters can be hormonally suppressed in the presence of their mothers has led to transfers of female offspring to other zoos, increasing their chance of breeding. Breeding with white rhinos is necessary for a stable, self-sustaining population. Even more, reproductive disorders are often an age related consequence of long non-reproductive periods (Hermes et all, 2007). Those reproductive disorders can compromise the well-being of white rhino severely. Non-breeding specimens should be assessed for reproductive pathologies. If a female is diagnosed with a reproductive pathology, it would not make sense to have her transferred to a different herd to stimulate sexual behaviour. The reproductive health status is a very important subject to be able to make any decision regarding these kind of animals, whether male or female.
In 2001 the EEP Species committee recommended that the birth interval for white rhinos should be no less than 4 years. For extensive groups the birth interval depended on the individual animal, because normally mother and calf are not separated (Goltenboth *et al.,* 2001). In a multi-male group, incomplete dominance of the males and interference from other males during mating can result in a reproductive failure. In such a case, the solution is to maintain one male with the females and separate the remainder as a bachelor herd (Seror *et al*., 2002).

2.5 Behavioural enrichment

Captive environments are considerably less complex than wild environments. Therefore, confined animals can show boredom in response to an enclosure that fails to stimulate their wild behavioural needs. It is difficult to define the stimulatory needs of an animal as they vary between species and even between individuals. To maintain natural behaviour in captivity, it is essential to adapt captive conditions to the animal with behavioural enrichment, rather than expecting the animal to adapt to the environment (Carlstead, 1996). This chapter provides an overview of possible methods of behavioural enrichment for captive white rhinos. The responses to the methods used for enrichment vary from individual to individual. Thus, the methods in this chapter are not a guarantee for success. It should also always be taken into consideration that new things can cause stress for the rhinos.

2.5.1 Rhino behaviour

Behavioural enrichment should suit the species specific needs and the physical build of the animals. As stated in § 1.1.3, white rhinos have strongly developed senses of smell and hearing (vocalisations are listed in table 11 below (Owen-Smith, 1973; Policht *et al.*, 2008)). These senses are most suitable for white rhino enrichment.

Table 11. ***White rhino calls***

Sounds	Call	Situation / meaning
Tonal sounds	whine*	begging for food
	squeak*	separation
Puffing sounds	snort*	no obvious
	threat	first warning
	puff	no obvious
Growling sounds	snarl*	aggressive
	grunt*	powerful warning
	grouch	foraging and other activities in proximity of other members of the herd
	groan	moan, body discomfort
Repetitive sounds	pant*	greeting, contact call
	hoarse	feeding, approach to female

*** = calls recorded also in Southern white rhinoceroses (Owen-Smith, 1973). Other calls of the Southern white rhino (“shriek, squeak, squeal, gruff squeal and gasp-puff”) were not recorded in this study of Northern white rhinoceroses.**

<u>Stress, aggression and stereotypic behaviour</u>

Animals in captivity can display stress, aggression and stereotypic behaviour. The goal of enrichment is to minimize these behaviours by stimulating species specific behaviours. Behaviours that indicate stress in white rhinos are listed in table 12 (below). Aggression can be a result of stress, table 13 below shows how to minimize stress. The stereotypic behaviour displayed by white rhinos are weaving, extensive horn rubbing, pacing, bar biting, licking metal barriers, rubbing, abrasion and lethargy. This can be caused by boredom, small enclosure size, unable to escape other group members, unable to display all the natural behaviours, stress caused by their environment (i.e. public, machinery) and loneliness. Refurnishing, changing contact moments (like variable feeding times, etc.), training, enrichment, mixing with other species and changing group composition can be used to minimize stereotypic behaviour.

Table 12. ***Stress indicators***

Stress indicators
Pacing
Change in normal behaviour
Running or increased locomotion
Spending time away from the herd
Aggression
Increased vocalisation and snorting
Loss of appetite
Reduced laying time
Unresponsive or quiet
Loose faeces and more frequent defecating

Table 13. ***How to minimize stress***

Minimize stress by
Enrichment
Providing enough space
Separate feedings
Individual pens

2.5.2 Types of enrichment

There are numerous ways to keep a white rhino occupied and encouraged to show its natural behaviours. The most effective enrichment types are food enrichment, scent enrichment, item enrichment and management (training, breeding, splitting up the herd).

Food enrichment

Food enrichment, when used, should be part of a balanced diet to keep track of all the intake of the individual animals. When using food as enrichment for white rhinos, it is important to keep in mind that food items used are taken off their daily diet allowance, so they do not get overfed. It is also good to make sure that no toxic plants or vegetables are fed.
The most suitable foods for enrichment are fruits, vegetables, pellets, hay or straw and bread. Other proven food enrichment is peanut butter, honey, pumpkins, watermelon, ice blocks (see figure 23), snow, corn syrup, molasses, alfalfa cubs and a non-regular diet (IRKA, 2010c). Check for toxicity §2.2.1.

Figure 23*. A white rhino at Marwell Wildlife Park, UK, is inspecting a blackcurrant ice lolly*

Figure 24. A white rhino in the Phoenix Zoo investigates the scented bowling balls

Scent enrichment

Hadley (2000) did a scent research by putting liquid scent extract on a sponge and putting this in a bowling ball (see figure 24). She found that Southern white rhinos have little interest in the almond, coconut, Old Spice, vanilla, and vinegar scents. Two scents (banana and maple) had more interactions, but peppermint had the longest duration and anise extract caused the highest total number of interactions. The responses to the scents included ignoring, looking near (within three feet), sniffing, rolling the bowling ball under the nose, rolling the bowling ball across the exhibit, mouthing the bowling ball, and marking (urinating on) the bowling ball. Hadley (2000) concluded that "any scent that is identified as favourite could be used in other forms of enrichment. For example, the scent could be placed on one or more of the logs, used to create a scent trail on the floor of the exhibit, or painted on the walls in the exhibit. This may result in more exhibit exploration by the rhino, as well as encouraging natural behaviours like patrolling and marking territory." She also noted increased interest by zoo visitors, so this provided an excellent opportunity to further educate the zoo public about the natural history of the white rhinoceros, and to explain the purpose of enrichment.

Burrell *et al.* (2004) did a research on dung enrichment in black rhinos. They used dung from conspecifics and other ungulate species. They found a significant difference between the frequency of faeces investigation on days with conspecific dung enrichment and baseline days. Other proven scent enrichment items are: spices and herbs, cinnamon on hay, urine and dung, pumpkin pies, scented oils, cologne, fur and pinecones (IRKA, 2010c). Not all rhinos respond to scent enrichment.

Item enrichment

Many different objects can be used as enrichment for white rhinos. Suitable items are logs, stumps, balls, kegs, rubbing posts, ponds and mud baths, sprinkles, loose, heavy chain fastened at both ends, logs suspended on chains and dung from other zoos. Other proven enrichment items are boomer-, root-, soccer-, bowling balls, traffic cones, kegs, wind chimes, hay bags, soap bubbles, leaf-, litter-, sand piles, spools, boat bumpers, car brushes, street brushes, hanging rubber mats, misters and showers, hanging hose, logs, sod, barrels, beer kegs, cardboard boxes, wheel barrow bucket, carpet tubes, Christmas trees and bark (IRKA, 2010c). Tires are not really suitable for the rhinos because they can get their horns stuck in them.

Stumps can be placed against the wall. This is a form of enrichment and it also improves the exterior of the stables for the visitors.

Management

Enrichment doesn't have to involve putting items in the enclosure with the animal. It can be effective just by breaking habits. Rotating yards, shifting to other stalls and howdy gates can be used as well as shifting rhinos into the exhibit overnight, training (see §2.6.2), variable feeding times, semi-tactile contact with other rhinos on a variable schedule, moving exhibit furniture, splitting up the herd and playing recorded audio clips of other rhinos. (IRKA, 2010c) It should be taken into account that this can also cause unnecessary stress. Rhinos are creatures of habit, so big changes can be very stressful for them.

2.5.3 Implementation

The power of enrichment lies in its novelty. Everything new is something to explore for the animal, but unfortunately, it works the other way around as well; the effect of enrichment fades away when enrichment is applied constantly or in a predictable setting. It is dependent on the type of enrichment and the available time of the keepers when enrichment should be available. It also varies from individual to individual. The institution can choose for natural enrichment or unnatural enrichment. Both are effective, but the strategy has to be in line with the institutions philosophy. Observation is always important to assess how effective the enrichment is and to see if the enrichment is safe.

Enrichment can be applied in a number of ways, for example by pulley systems, ropes, in water, cables across the yard, chains, browse holders, bait tree, logs with holes drilled in it, buried, tire totem, scattered, piñatas, hay racks and rubbed on to things.

Regarding safety, things like sharp edges, broken zip lines, items suspended too low, throwing or launching items, chemical treated wood, breakage of ceiling lights, sky lights, ceiling fans, entanglement, barrier problems and impact on hot wire, food toxicity, door obstruction by free rolling items and water (seasonal indications) should be kept in mind. With regard to the safety of the animal it’s important that the animal cannot injure itself or other animals. (IRKA, 2010c)

2.6 Handling

This chapter contains information about identification, training, transport and safety. When keeping white rhinos, the keepers should be able to tell the rhinos apart. This is useful for management and/or medical reasons. When transporting a white rhino, the stress levels should be kept as low as possible. By training the rhino, the animals can get familiar with the crate and the procedure of getting ready for transport. Special adjustments must be made to create a safe working space for the keepers and safe enclosures for the white rhinos. Also the possibility of an escape is discussed.

2.6.1 Individual identification and sexing

The best way to identify white rhinos is just to get to know their external features. A microchip is recommended as officially recognised identification. Microchip brands that are used are: paddy-mark and any ISO chip used in the EU.

The best time to chip a calf is with the first handling or in the first week of its life, when it's still hand manageable. The best place to chip the rhino is around the neck or shoulder, because there are a lot of muscles there. The base of the ear of tail is also possible, but this is much more sensitive.

2.6.2 General handling

It is advisable to train the rhinos for inspections and treatments. This should always be done with a barrier between keeper and animal. Holden *et al* (2006) found that operant-conditioning can be an important aspect of the daily husbandry routine. By training the rhinos the management and monitoring of the rhinos can be carried out more effectively and with minimal stress and risks for the animals and keepers. Numerous procedures can be carried out without the need for immobilization and the trust between the animals and the keeper and veterinary staff will grow.
The program can also be seen as a form of stimulation for rhinos in captivity, improving physical and psychological well-being and complementing other aspects of husbandry, such as environmental enrichment and habitat complexity. A list of commands and training tips of the International Rhino Keepers Association (IRKA 2010b) can be found in appendix II.

2.6.3 Catching/restraining

Except for young calves, physical restraint of rhinos is next to impossible. The nearest approach to physical restraint is to entice the animal into a shipping crate/shute with bars, through which a handler can carry out gentle manipulations (Fowler 1995).
The doors of the pens and stables can be closed hydraulically or manually to prevent accidents.
It is advised to include a shute in the stables. This way the rhinos can be easily guided into the transport box when needed. A squeeze cage can also be helpful in the stable for medical treatments and safety reasons.
In general, it is highly recommended that institutions modifying rhino exhibits or constructing new ones incorporate a physical restraint area or device into their design considerations. Several physical restraint designs are effective for rhinos. In general, major restraint chute design considerations include strength, durability, type and function. It should be noted, that available space and animal's size and disposition vary across institutions and should be individually addressed. A restraint chute or restraint area can be designed so that the rhinos must pass through it to exit the barn into the yard. If rhinos are fed indoors, part of the feed can be offered in the chute area. Rhino chutes should be manufactured out of steel or a combination of steel and steel-reinforced wood. Steel-strength aluminium has also been used. Aluminium is lighter and more manoeuvrable than steel, as well as potentially less stressful to rhinos because of lower sound properties than steel (Fouraker and Wagener, 1996). It is important to give the rhino time to get used to the chute so that it is calm and relaxed while being restrained. Depending on the temperament of the individual this may take many months to accomplish.

Restraint chutes: Permanent pass-through indoor restraint chutes are especially effective for rhinos. The chute should allow restraint of the animals when it is passing through in either direction so that shifting routine of the animal is not interrupted. The width of the chute should limit side-to-side movement while still allowing the animal to comfortably lie down. However, animals can become wedged in tight-fitting chutes if the side cannot be released. To alleviate excessive forward movement of the animal when it lowers its head, two vertical bars that push in from sides of the chute to the shoulder of the rhino may be utilised. Quick release of these shoulder bars often relieves agitated animals without having to release them completely (Fouraker and Wagener, 1996).

High-walled chutes: High-walled chutes or bars over the top keep the animals from climbing or rearing up. Horizontal bars in the chute's entry gates and sides are hazardous for examiners when the animal lies down. Vertical bars on the sides can trap researchers' arms if the animal can move forward. If the animal's movement forward and side-to-side mobility can be limited, vertical bars or walls on all sides are recommended. The distance between these bars along the sides of the chute should be great enough to prevent the animal's foot from becoming wedged if the animal rolls on its side in the chute. For researcher safety, this distance can be divided with removable vertical bars (Fouraker and Wagener, 1996).

Closed chute: A closed chute is another option that has been used successfully. A typical closed chute has both front and back gates. The back gate restricts the rhino's movement by sliding forward. The hind end of the rhino is supported by a v-design that prevents it from lying down. This design also allows additional safety for the staff while working with the animal. In many respects, a closed chute does not depend as strongly on conditioning of the rhinos as does a squeeze chute, though acclimation is recommended prior to attempting any treatments within the chute. The design of a closed chute might necessitate an outdoor location in most cases, thus the use of this type of chute may be limited by weather (Fouraker and Wagener, 1996).

Free-stall chute: A free-stall chute can be used for animals more sensitive to a confined enclosure. The design of this type of chute allows the rhino to enter or exit at its will and thus may help to keep rhinos calmer during procedures. Because there is free access rhinos must be conditioned to target or stand still. A free-stall design can easily be incorporated into an existing pen or stall, indoor or outdoor. As stated, the open back of this type of chute allows the animal to enter and leave the structure at will. Protection of staff when working with the rhino is important, and a partial back wall constructed of vertical pipes allows staff to step out of the way (Fouraker and Wagener, 1996).

2.6.4 Transport

The IATA guidelines for transport apply when a white rhino is being transported. These guidelines give criteria for the crate and the care of the animal during transport. The IATA guidelines for transport should be followed when an animal is transported to another holding facility. For transportation, loading rhinos into a trailer has worked, but crating is preferred. First step will be to introduce the crate as a none-interactive part of the environment, which is followed by placing food into the crate. For training, the crate can be placed between the indoor and outdoor enclosure. This way the animal will get accustomed to passing through it (Goltenboth *et al*., 2001). The animal gets used to being in the crate and it won't stress him out when transported. Though it should be taken into consideration that the animal can be very scared of the crate and won't come inside anymore. The animal's overall condition should be closely monitored when transported on a trailer. Forced crating should not be practiced (Goltenboth *et al*., 2001).

Container/ crate

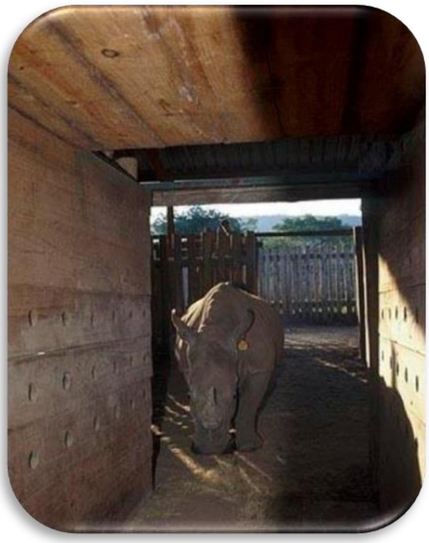

Figure 25. Rhino walking into crate

During the transport the crate shown below (figure 26) or similar should be used. Larger crates are normally made of wood, reinforced with steel. (see figure 25) The crate's size should be designed in accordance to the animal's volume; it should always be larger in size than the animal by 1 m in width and length when laying on its sternum, but the rhino must not be able to turn.
Narrow bars at the front helps preventing eye and face injuries. To prevent damage of the horns or facial injuries, the bars should be angled away from the animal. Sharp edges should be paid special attention to (Goltenboth *et al.*, 2001). As soon as the rhino is in the crate, two bars should be pushed in the crate horizontally near the front/back of the crate. Also a small hatch to feed and inspect the animal is recommended. In general an iron frame should be used in the whole crate, with the wood as a second barrier.

In preparation of the transport the animals should get a relaxation agent. Perphenazine/ trilafon can have a positive effect in many species, because it keeps them calm even during the introduction. Crate training will also calm the animal. Tranquilizing can be dangerous since the animal could hurt itself. The best option to keep the white rhino as calm and stress free as possible is to start crate training from one month before the transport (Goltenboth *et al.*, 2001).

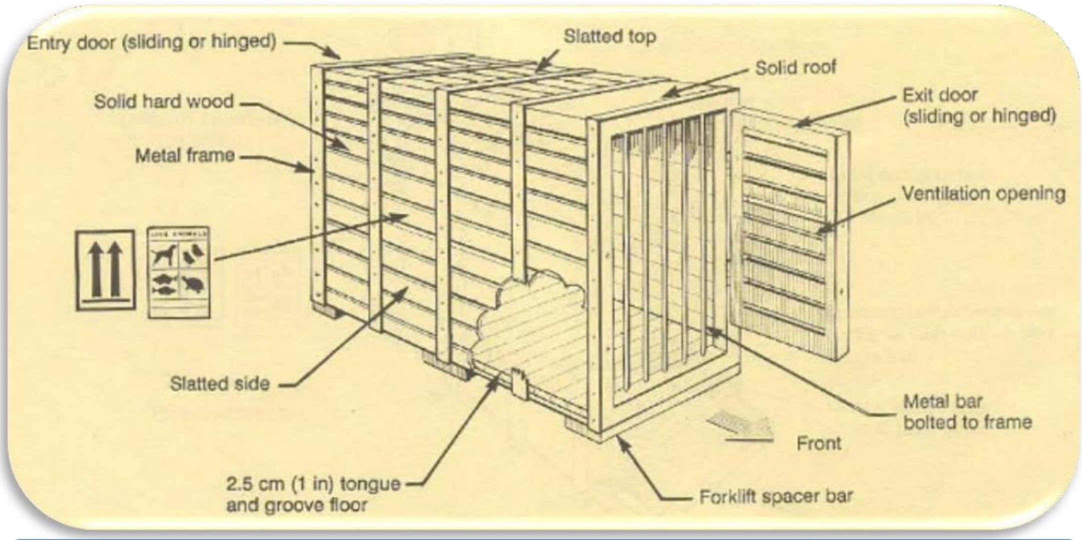

Figure 26. IATA crate

2.6.5 Safety

Metal edges, nuts and screws must be secured with wood fittings to prevent injuries to the animals and staff. It is important to always consider fence spacing and keeper access/exit in case of emergency. Keepers should always fit through any two bars of the fence (Goltenboth *et al*., 2001).

The following regulations should be kept in mind at all times when working with white rhinos:

- No direct contact with the animals, unless absolutely necessary
- Always be alert and don’t make sudden noises or movements that can startle the animal
- Always look for escape passages
- Watch your feet

Due to training the white rhino, it can easily be examined with the least stress and highest safety levels for the keepers and vets (Goltenboth *et al*., 2001).

Escape

When a white rhino escapes, the best way to handle is to evacuate the visitors out of the park or distance and then try to lure the rhino back to its enclosure with food. If this doesn’t work, the animal can be closed in with trucks to persuade it to go to the enclosure on its own. Another option is to tranquilize the animal and then take it back to the enclosure. If all fails and there is a high security risk, there should be a powerful enough firearm at hand.

2.7 Veterinary

This section briefly outlines any physical conditions or complaints commonly associated with white rhinos. Symptoms, treatment and prevention of common diseases and conditions are outlined. Required vaccinations are specified. Also common parasites, screening and treatments are described as well as information on causes of mortality. For all diseases and injuries the adagio that "prevention is better than cure" is the way to cope with Good Veterinary Practice (GVP).

The behavioural repertoire of rhinos is quite limited regarding illness. Depression and inappetence are often the only signs of major disease problems (Fouraker and Wagener, 1996). Described diseases and injuries of white rhinos in captivity are skin lesions, gastrointestinal torsion and impaction (resulting in severe colic), leiomyomas, pleuritis, pneumonia, clostridium infections, fertility problems, internal tumours, pododermatitis and tuberculosis. Dental problems are often a problem at very old age, resulting in emaciation and death. In table 14 (below) the inoculations are listed. White rhinos should be trained to ease inspection and treatment (see figure 27 and §2.6.2).

Figure 27. Keepers are inspecting a Southern white rhino

Table 14. ***White rhino inoculations with time interval***

Type of inoculations	Time interval
Deworming	Twice per year depending on faecal parasitological tests.
Tetanus (depending on region)	Once per year.
Vaccination against clostridium	Once per year. After initial vaccination, booster 1 month later.

2.7.1 EAZA Bio Bank

There is a call from the EAZA BioBank to collect extra blood, tissue and/or serum samples whenever an animal is sedated. This helps supporting population management and conservation research. The sampling protocol can be found in Appendix III.

2.7.2 Foot problems

Nail cracks

The most common problem seen in all rhino species is vertical cracking in the nail wall, which can range from mild quarter cracks to more extensive splitting up to the corona. Trauma to the bottom of the nail or the coronary band can generate cracks and concrete flooring aggravates this by wearing and thinning the nail walls of the lateral toes while the rhino is lying down. Straw can make a hard concrete floor feel softer. Hooves have a natural waterproof, external layer called the periople, which provides a protective coating and regulates evaporation and absorption. The periople can be damaged by concrete, sandy soils, chemicals or improper filing. Dry, brittle nails lose resiliency and are more prone to splitting. Excess moisture can also cause damage. This moisture balance is influenced by the external environment or affected by an inappropriate diet. Less prevalent are horizontal cracks in the nail, which may occur after a serious illness, laminitis or nutritional disorder (Jacobsen, 2002).

Treatment for cracked nails starts with cleaning the foot and carefully removing mud, grit or faeces. This allows for closer inspection and keeps debris from wedging and opening the crack further. Topical antiseptics may be prescribed to prevent infection. Commercial hoof dressings should only be used with veterinary approval since some of these products contain turpentine or petroleum compounds and the splitting nails may not be caused by dryness. Corrective trimming by experienced personnel can be used to relieve pressure on the bottom of the nail and enable the crack to grow out. However, by making changes in environment and husbandry, most of these cracks can be allowed to grow out without intervention (Jacobsen, 2002).

Laminitis

Also referred to as founder, laminitis is a metabolic and vascular disease which can affect rhinos and other hoof stock. The disease starts when the blood supply to the corium (the sensitive laminae of the foot) is interrupted. Damage to the coronary corium causes band or irregular horn growth called laminitic rings. In severe cases, the union between the horny and sensitive laminae breaks down and progresses to separation of the nail at the coronary band. Some common causes of this disease are excessive feeding of concentrates, enteritis, chronic renal failure, and IHVS (idiopathic haemorrhagic vasculopathy syndrome) (Jacobsen, 2002).

The first signs of laminitis are lameness and inflammation or discharge at the coronary band. Gradually a gap appears at the top of the nail. It is possible for the affected nail to remain while the new nail grows and replaces it. With total separation, the nail is only attached at the sole and tends to fold under the foot as the rhino walks. In this case, the nail is removed under anaesthesia. Post-op treatments include good hygiene, keeping the foot and exposed laminae clean, topical antiseptics and pain management. New nail growth is usually completed in six months (Jacobsen, 2002).

2.7.3 Skin problems

Described skin problems in white rhinoceroses often reflect to Malassezia dermatitis, a yeast infection of the skin (de Meurrichy, 1996). Topical treatment including antifungals will result in improvement of the condition.

2.7.4 Parasites

Parasites only play a major role in newly wild caught animals and in animals kept on grass land in tropical climatic zones. With the modern broad range anthelmintics for horses, also rhinos can be treated (according to body weight) successfully and without problems. If an animal has newly arrived from an area of tropical climate, screening should include blood exams for haemic parasites, trypanosomiasis, theileriasis and leishmaniasis (Goltenboth *et al.,* 2001). In table 15 (Goltenboth *et al.,* 2001) descriptions and treatments are given for parasites.

Table 15. ***Parasites, description and treatment in white rhinos***

Parasites	Description	Treatment
Protozoa	**Trypanosoma, Babesia, Theileria are wide-spread in rhinos in Africa but are of no relevance in zoos.**	**Treatment has been tried with toltrazuril (Baycox bovis) or amprolium**
Gastro-intestinal protozoa	**Coccidiosis, Balantidium coli and Trichomoniasis occasionally cause diarrhea especially in young rhinoceroses.**	**Successful treatment has been executed with Baycox bovis (toltrazuril), amprolium (AmprolvetR, MSD-Agvet) and jodochlor hydro-xyquin.**
Cestodes	**Rhinos, newly arrived from the wild, for years may show proglottides in the faeces from**	**Recently successful treatment has been executed with**

	Anoplocephala spp. and A. gigantea. Animals weakened by other illnesses frequently show massive infestation.	praziquantel at a dose of 0,5 - 1 mg/kg BW.
Nematodes	A large number of gastro-intestinal nematodes has been seen, such as Strongyloides sp., Kiluluma sp., Quilonia sp., Drascheia sp., Probstmayria sp., Oxyuris karamoja, Habronema khalili, Parabronoma rhinocerotis, Grammocephalus intermedius and Gr. clathrotus.	The application of modern broad range anthelmintics such as thiabendazole, fenbendazole, mebendazole, fenbendazole, albendazole, pyranthel tartrate and ivermectin keep parasites well under control (doses to be based on those for horses).
Arthropodes	White rhinos are often carrier of larvae of the warble-fly Gyrostigma pavesii. Also, Gastrophilus-larvae are frequently found in rhinos newly arriving from the wild. The larvae cling to the gastric wall and are excreted with the faeces. Severe infestation can cause inflammations. In imported rhinos, for some time flies may still hatch from larvae when kept in well heated enclosures.	Treatment with ivermectin per os will eliminate the larvae.

Parasites have been found on low frequency and are usually not associated with clinical signs in captive white rhinos (Fouraker and Wagener, 1996). The parasites that are found are listed in table 16.

Table 16. ***Parasites found on white rhinos in captivity***

Type of parasite	Parasite
Ecto-parasites	ticks
	mites
	flies
	ear mites, fly strike i.e. maggots around the horn base
Endo-parasites	tapeworms
	stomach botfly larvae
	nematodes

2.7.5 Bacterial infections

Pressure sores and ulcers are common, especially in older animals and are a porte d'entreé for bacterial infections. Multi-bacterial infections can be hazardous to all rhino species. They are caused mainly by traumatic injuries, affection of the lungs and the gastro-intestinal tract and they often lead to general septicaemia that is proven to be fatal. Successful treatment is dependent on the possibility to apply high doses of the appropriate antibiotics.

It must be stressed that tuberculosis is dangerous to all rhino species. Like in other zoo animals, intracutaneous tuberculinisation is best set on the upper eyelid. Set behind the ear or on the tail is less reliable. Supportive ante mortem tests include ELISA-tests, MAPIA test, etc.(Miller, 2008) or the direct demonstration of the infectious agent at post mortem. Treatment is very rarely indicated and seldom shows satisfactory results. Salmonella is one of the most frequently reported bacterial pathogens in rhinoceroses (Ramsey, 1993). Salmonella-infections can successfully be treated with enrofloxacin (BaytrilR, Bayer) (Goltenboth *et al.,* 2001).

2.7.6 Viral diseases

Poxvirus infections have been described in both black and white rhinoceroses, showing acute systemic disturbances. Lesions include vesicles and pustules of various sizes on the flank, abdomen and medial thigh. Ulcerations may also be present at the mucocutaneous junctions and the mucosa of the oesophagus and stomach (Ramsey 1993). Vaccination against elephant-pox is recommended with the Modified Vaccinia-Virus Ankara (MVA). In cases of ulcerative dermatitis and pox-like skin diseases, the presence of a Herpesvirus-infection should always be taken into consideration (Goltenboth *et al.,* 2001).

2.7.7 Reproductive disorders

The incidence of reproductive disorders is greater in nulliparous females and is positively correlated with age. Reproductive disorders are regarded as age-related consequences of long non-reproductive periods (Hermes and Hildebrand, 2011).

Males

Several diseases of the male reproductive tract have been described, including trauma to the penis, diseases of the accessory sex glands and testicular fibrosis, atrophy, trauma or neoplasia and epididymidal cysts (Hermes and Hildebrand, 2011).

Females

Anoestrous is a common finding in white rhinos and is considered as a primary cause for the low reproductive rate in captivity. Endocrine data, ultrasound and post-mortem findings have revealed that anoestrous is associated with different status of ovarian activity. Young females in anoestrous exhibit high ovarian activity and regular follicular waves. However, ovulatory-sized follicles do not ovulate but become atretic or haemorrhagic (Hermes and Hildebrand, 2011).
Anecdotal reports that the transport of females to other facilities or the introduction of new males initiated regular oestrous cycle activity, indicate that the impact of behavioural aspects to initiate or resume the oestrous cycle activity are underestimated. All scientific data support the conclusion that anoestrous in young females is primarily the result of deficient animal husbandry/management, which fails to provide the behavioural needs to initiate the regular oestrous cycle activity. Over decades, this ultimately leads to depletion of oocytes, resulting in a secondary cessation of reproductive activity and premature senescence in mid-aged females (Hermes et al. 2007).
Early embryonic loss may be associated with uterine inflammation or pyometra in individual cases, but is clinically uneventful. Luteal insufficiency has been a suspected cause in animals with a history of embryonic resorption in captivity (Hermes and Hildebrand, 2011). Dystocia and stillbirth occur in all captive rhinoceros species. Early detachment of the placenta, malposition or fetal malformation are incidental causes reported for stillbirth (Hermes and Hildebrand, 2011).
The increased use of ultrasound has facilitated in vivo diagnosis of reproductive disorders and evaluation of reproductive potential in rhinoceroses. In general, reproductive pathologies in female rhinoceroses involve ovarian, uterine, cervical and vaginal tumours, endometrial and ovarian cysts, endometrial hyperplasia and mucohydrometra (Hermes and Hildebrand, 2011). Cystic or neoplastic formations can be a hidden source of discomfort, preventing intromission, mating and/or semen transport. The incidence of reproductive disorders is greater in nulliparous females and is positively

correlated with age (Hermes and Hildebrand, 2011). Reproductive disorders, along with ovarian exhaustion, might render a female irreversibly infertile early during life. This phenomenon has been termed "asymmetrical reproductive aging" (Hermes and Hildebrand, 2011)(Hermes *et all*, 2006).

2.7.8 Mortality

The most common causes of mortality of adult rhinos in captivity are old age, fighting, diseases and accidents. In juveniles fighting and diseases are the most common causes of captive mortality. To be absolutely sure about the cause of mortality, a post mortem investigation should be done on every deceased white rhino. Therefore, an institution holding white rhinos should have a post mortem protocol.

2.7.9 Diet

Nordstrom and Bissonette (2006) found that "the frequency of health problems was mainly determined by diet, with the most problems occurring when pelleted feed was more than 40% of the total diet. Skin problems, although a rare occurrence, were the most frequently reported health problems. Climate also may play a role in the frequency of health problems, with more health problems recorded in zoos in warm, dry climates, but less skin problems in warm, wet climates." Other dietary problems are listed in table 17 (Clauss and Hatt, 2006).

Table 17. ***Diet based health problems***

Health problem	Recommendation
Farmer's lung condition	**Hay should account for the major proportion of any diet for white rhinoceroses in captivity. The importance of the hygienic quality of the hay has been emphasized by cases resembling a "farmer's lung condition"**
Hypophosphataemia	**Roughage based diets are particularly vulnerable to phosphorous deficiency. Hypophosphataemia (low levels of phosphorus in the blood) has been observed in rhinoceros with haemolytic crises, so a deficiency of this mineral in the diet should be avoided.**
Colic	**Avoid ingestion of sand, which can cause colic.**
Skin and foot lesions	**Zinc deficiency may lead to the development of skin and foot lesions.**
Skin and eye diseases and disturbances of the digestive system	**Vitamin A deficiency, high doses of vitamin A should be applied intramuscular (Goltenboth *et al.*, 2001).**

2.7.10 Anaesthetics

"Due to the size and behaviour, anaesthetic management of white rhinoceroses is challenging and adequate planning is necessary to minimize the risks (Raath, 1999). Injectable anaesthetic techniques are more frequently used than inhalant anaesthesia, since most procedures are short and performed under field conditions. However, for longer recumbency times or more invasive surgical procedures, the use of inhalant anaesthesia is preferred." (Valverde *et al.*, 2010) See §2.6.3 for the right enclosure design for optimal use when catching or restraining a white rhino.
Many drug combinations have been used successfully in the anaesthesia of white rhinoceroses (Raath 1999).
Immobilon (2,25 mg/ml etorphine + acepromazine) on its own has successfully been used in the past in small dosages (up to 1.6 ml). Hyperthermia and a rather bad induction (muscle tremors) makes Immobilon obsolete. The effects are a lot less prominent in combination with detomidine. For a

female white rhino 3 mg etorphine + 12 mg detomidine is recommended by Kreeger et al. and for a male white rhino 4 mg etorphine in combination with 20 mg detomidine is recommended.
Nowadays a combination of butorphanol (20-30 mg) and detomidine (20-30 mg) i.m. is widely used in adult animals. Addition of ketamin (100 -150 mg) i.v. can be used to prolong or deepen the anaesthesia. Even Immobilon can be added for more painful interactions, in very small amounts such as 0.2 ml.
All additional drugs, such as emergency drugs (antidotes, antibiotics) and tranquilisers, must be ready and checked before darting commences. Provisions must include a kit to treat accidental human injection with opioids (Raath 1999).
Antidotes commonly used to reverse rhinoceros anaesthesia include the agonistic antagonists nalorphine and diprenorphine hydrochloride (M5050, C-vet) and the pure antagonists naloxone (Narcan, Boots) and Naltrexone (Raath 1999) against opiods. When using dipherenorphine renarcotisation often occurs. This can be avoided by using naltrexone, which is a drug preferred to antagonise opioid drugs. Atipamazole is the drug of choice to antagonise alpha 2 adrenergic drugs such as detomidine, medetomidine and xylazin.
A well-vascularised muscle area should be selected for the dart placement. The darts must be placed at right angles to the skin to avoid subcutaneous deposition of the drug (Raath 1999). With low doses, the lips provide a perfectly well vascularised area to inject anaesthetics.
The use of long-acting tranquillizers for transports and introductions in other facilities either the short acting zuclopentixol (adults 300 mg) or the longer acting perphenazine enanthate (adults 300 mg, juveniles 100 mg) can be used safely.

2.8 Specific problems

As said before, numbers of white rhinos in the wild are increasing, but the European population in captivity is not self-sustaining. This can have several causes: only a small percentage of the imported animals from the 1970s have reproduced themselves, animals start breeding too old, animals do not have a cycle or animals do not conceive. Especially the second generation captive individuals have bred poorly. One hypothesis is that young animals need to be transferred out of their maternal herd once they are juvenile, as well as adult females that do not conceive or do not have a cycle. A transfer to a different location and herd may affect their reproductive biology in such a way that they will become successful breeders.
The EEP is convinced that introductions are a vital part of a successful breeding program. More attention should be given to introductions of individuals into groups, space for (temporarily) separation and space for retreat of individuals during introductions.
Female white rhinos in the wild may live in herds of approximately six animals. Males live solitary and they only meet females to breed. This is hard to realize in captivity because of the big enclosure size needed. It should be prevented that white rhinos are held isolated or kept in small exhibits and one should try to mimic the natural ecology as much as possible.

The existence of these problems shows that not all is known about keeping white rhinos. This is why more research needs to be done and the husbandry guidelines need to be updated regularly. These husbandry guidelines are one step forward to an uniform best-practice policy in EAZA institutions.

2.9 Recommended research

This is a list of possible interesting additional research subjects:

- Introduction techniques for calves to males
- More research on Tb testing
- Sex of offspring relative to quality of food
- Social structure and (female) group composition in correlation with breeding results
- Hormonal deprivation of young females
- General research on captive breeding
- Further research into copper metabolism. (Clauss and Hatt, 2006)

3. Discussion

EAZA is developing Best Practice guidelines to stimulate higher welfare in member institutions, aiming for better reproduction success and easier exchange of animals between EAZA institutions because of uniform conditions. Proper animal husbandry is needed for good population management and helps conservation of the white rhinoceros. (EAZA, 2018a)

Within the white rhino EEP opinions on some topics, like social structure and especially breeding, differ greatly. This is partly due to personal beliefs, as in many cases it is unknown why certain problems arise. Are Safari exhibits more successful then zoo exhibits when it comes to breeding with white rhinos? Is the welfare of white rhinos compromised in small exhibits? These are subjects which continuously cause discussion within the white rhino EEP. Female juvenile rhinos in the wild disperse to live with other (unrelated) rhinos many times, but not always. Do we necessarily need to transfer our female juvenile rhinos? There have been examples of rhinos which had reproductive problems (cycle problems, conceiving problems) and which without difficulty conceived and gave birth after a transfer, but is this the "golden rule"? The signals of the last 10 years surely point into this direction with the increase of natural pregnancies in Captive born rhino.
In the wild, females group up to six animals and males live basically solitary and associate only with females in oestrus (Tomasova, 2006). In captivity it is advised to hold 2.3 white rhinos, but only if the bulls can be provided with separate territories. This has implications on enclosure size and design when wanting to mimic the wild situation.
A white rhino in the wild is consuming large amounts of short grasses (Steuer et al., 2010). The captive situation is in great contrast with the natural feeding ecology. Zoos are still feeding fruits, vegetables and processed food like bread and flaked maize in addition to grass and hay.

These are all examples of subjects of which we just don't know enough. Continuous research and attention should be given to this magnificent species, to improve even further our historical bad breeding track record. The last 10 years shows that we are on the right track here! With the enormous poaching pressure in the native range countries we are obligated to do so. There are famous examples where captive populations have saved a species from extinction. We need to be ready to do this if ever this species becomes nearly extinct. Remember, in the beginning of 1900 we already almost lost this species to extinction.

4. References

4.1 Books

Baarda, D.B. and Goede, M.P.M de, 2001, *Basisboek Methoden en Technieken: Handleiding voor het opzetten en uitvoeren van onderzoek*. Wolters-Noordhoff, Groningen, Netherlands

Carlstead, K. 1996, *Effects of captivity on the behaviours of wild mammals.* in: Wild Mammals in Captivity. Chicago University Press, Chicago, USA

Emslie, R. and Brooks, M.1999, *African Rhino Status Survey and Conservation Action Plan*, IUCN/SSC African Rhino Specialist group. IUCN , Gland, Switzerland and Cambridge, UK

Fowler E. 1995, *Restraint and handling of wild and domestic animals.* Iowa state university press. Ames, USA

Hosey, G., Melfi, V. and Pankhurst, S. 2009, *Zoo Animals, Behaviour, Management and Welfare*. Oxford University Press. Oxford, UK

Hermes R., Hildebrandt T.B. 2011. *Theriogenology in rhinoceroses*. In: Fowler's Zoo and Wild Animal Medicine, current therapy 7, Eds. Fowler, M.E. and Miller, R.E.,

Elsevier/Saunders, St. Louis, USA, p.p. 546-651

Kaandorp, J., The use of neuroleptic tranquilizers in zoological medicine, Verh.ber. Erkrg. Zootiere, (2005) 42, p.p. 16-26

Kingdon, J. 1997, *The Kingdon field guide to African mammals*. Academic Press, London

Kreeger, T.J., 2002, Handbook of Wildife Chemical Immobilization, international edition, Wildlife Pharmaceuticals, Fort Collins, USA, p.p. 245

McKenzie A.A. 1993. The capture and care manual. Wildlife Decision Support Services and the South African Veterinary Foundation, South Africa, p.p. 512-569

Miller M.A. 2008. *Current diagnostic methods for tuberculosis in zoo animals.* In: Fowler's Zoo and Wild Animal Medicine, current therapy 6, Eds. Fowler, M.E. and Miller,R.E., Elsevier/Saunders, St. Louis, USA, p.p. 10-19

Owen-Smith R.N. 1988, *Megaherbivores*. Cambridge University Press, Cambridge

Raath J.P. 1999. *Anesthesia of White rhinoceroses.* In: Fowler's Zoo and Wild Animal Medicine, current therapy 4, Eds. Fowler, M.E. and Miller, R.E., Saunders, Philadelphia, USA, p.p. 556-561

Ramsey E.C. 1993. *Infectious diseases of the rhinoceros and tapir*. In: Fowler's Zoo and Wild Animal Medicine, current therapy 3, Eds. Fowler, M.E., Saunders, Philadelphia, USA, p.p. 459-466

Stevens, C.E. and Hume, I.D. 1995, *Comparative physiology of the vertebrate digestive system*. Cambridge University Press, Cambridge, UK

4.2 Journals

Burchell, W.J. (1817) 'Note sur une nouvelle espece de Rhinoceros'. Bulletin des Sciences, par la Societe Philomatique de Paris. 1817, pp 96-97

Burrell, K., Wehnelt, S. and Waran, N. (2004) 'Olfactory enrichment and visitor effects in black rhinoceroses (*Diceros bicornis*) at two UK zoos'. Proceedings of the Annual Symposium on Zoo Research. 6, pp 187-201

Citino, S.B. and Bush, M., (2007) 'Reference cardiopulmonary physiologic parameters for standing, unrestrained white rhinoceroses (*Ceratotherium simum*)'. Journal of Zoo and Wildlife Medicine. 38.3, pp 375-379

Claus, M. and Hatt, J.M. (2006) 'The feeding of rhinoceros in captivity'. International Zoo Yearbook. 40, pp 197-206

Dierenfeld, E. S. (1999) 'Rhinoceros feeding and nutrition'. Zoo and wild animal medicine. 4 pp 568–571

Fouraker, M. and Wagener, T. (1996) 'AZA Rhinoceros Husbandry Resource Manual'. Fort Worth Zoological Park. pp 1-67

Goltenboth, R., Ganslosser, U. and Tomasova, K. (2001) 'African Rhinoceroses Husbandry Guidelines for Rhinoceroses'. International Studbook. 6

Griffith, B., Scott J.M, Carpenter J.W. and Reed. C. 1989. 'Translocation as a species conservation tool: status and strategy'. Science, New York 245, pp 477-480

Hadley, K. (2000) 'Scent Preferences of Southern white rhinos'. Shape of enrichment. 9.4, pp 1-3

Hermes, R., Hildebrand, T.B., Walzer, C., Göritz, F., Patton, M.L., Silinski, S., Anderson, M.J., Reid, C.E., Wibbelt, G., Tomasova, K. And Schwarzenberger, F. (2006) 'The effect of long non-reproductive periods on the genital health in captive female white rhinoceroces' Theriogenology 65, pp 1492-1515

Hermes R., Goeritz F., Streich W.J., Hildebrandt T.B. 2007. Assisted Reproduction in Female Rhinoceros and Elephants – Current Status and Future Perspective. Reprod Dom Anim 41, 1–12 (2007).

Hermes, R., Göritz, F., Saragusty, J., Sós, E., Molnar, V., Reid, C.E., Schwarzenberger, F. and Hildebrandt, T.B.(2008) 'First successful artificial insemination with frozen-thawed semen in rhinoceros' Theriogenology 71, pp 393–399

Hillman-Smith, K., Owen-Smith, R.N., Anderson, J.L., Hall-Martin, A.J.,and Selaladi, J.P. (1986) 'Age estimation of the white rhinoceros (*Ceratotherium simum*)'. Journal of Zoology. 210, pp 355-379

Holden, M.D., Gregory, J., Watkins, V. and Radfort, L., (2006) Operant-conditioning programme for white rhinoceros, Black rhinoceros and Indian or Greater one-horned Asian rhinoceros. Zoological Society of London, Whipsnade Wild Animal Park, Whipsnade.

Houssaye, F. and Budd, J.E. (2009) 'EAZA Husbandry Guidelines for the Leopard *Panthera pardus* spp'. European Association of Zoos and Aquaria (EAZA) felid tag. Amsterdam, Netherlands

Jacobsen, J., 2002. 'A review of rhino foot problems' pp 56-59
In: Aubery, L., Kennedy, J., Gaffney, M., Patton, L., Slobig, C., and Mehrdadfar, F., Proceedings of the [second] Rhino Keepers' Workshop 2001, Zoological Society of San Diego May 7-10, 2001. San Diego, Zoological Society

Jones, D.B., (1976) 'A bibliography of the land mammals of Southeast Asia' Department of Entomology. Taipei, Taiwan, pp. 1-238

Krelekamp, C. J. (2004) 'Husbandry guidelines Eurasian lynx *Lynx lynx* sspp'. European Association of Zoos and Aquaria (EAZA). Amsterdam, Netherlands

Milliken, T., Emslie, R.H. and Talukdar, B. (2017). African and Asian Rhinoceroses – Status, Conservation and Trade. CoP17, Doc. 68

Meurrichy de W., Bauwens, L., and De Vroey, C., (1996) 'A case of exfoliative dermatitis in a captive Southern white rhinoceros'. Journal of Zoo and Wildlife Magazine vol. 27, No 2, p.p. 271-274

Owen-Smith, R.N. (1973) 'The Behavioural Ecology of the white rhinoceros', University of Wisconsin

Patton, M.L., Swaisgood, R.R., Czekala, N.M., White, A.M., Fetter, G.A., Montagne, J.P., Rieches, R.G. and Lance, V.A. (1999) 'Reproductive cycle length and pregnancy in the Southern white rhinoceros (*Ceratotherium simum simum*) as determined by fecal pregnane analysis and observations of mating behavior'. Zoo Biology. 18.2, pp 111-127

Pienaar, D.J. (1994) 'Social organisation and behaviour of the white rhino' Proceedings of a symposium on"rhinos as game ranch animals", Onderstepoort, South Africa

Policht, R., Tomasova, K., Holeckova, D. and Frynta, D. (2008) 'The vocal repertoire in Northern white rhinoceros (*Ceratotherium simum cottoni*) as recorded in the last surviving herd'. Bioacoustics. 18, pp 69–96

Radcliffe, R.W., Farrel, S.T. and Chlids, S.E. (2000) 'Butorphanol and azaperone as a safe alternative for repeated chemical restraint in captive white rhinoceros (*Ceratotherium simum*)'. Journal of Zoo and Wildlife Medicine. 31.2, pp 196-200

Rosenthal, M. and Xanten, W.A. (1996) 'Structural and keeper consideration in exhibit design' Wild mammals in captivity: principles and techniques. pp 334-343

Rookmaaker, L.C. (2003) 'Why the name of the white rhinoceros is not appropriate'. Pachyderm. 34, pp 88-93

Schmidt, C. and Sachser, N. (1996) 'Auswirkungen unterschiedlicher Futterverteilungen auf Verhalten und Stresshormonkonzentrationen von Breitmaulnashornern (Ceratotherium simum simum) im Allwetterzoo Munster' Verhandlungen der Deutsche Zoologische Gesellschaft. 89.1, pp 294

Seal, U.S., Barton, R., Mather, L. and Gray, C. (1976) ‘Baseline laboratory data for the white rhinoceros (Ceratotherium simum simum)’. Journal of Zoo Animal Medicine. 7.1, pp 11-16

Seror, B., Zorovsky, Y., Terkel, A. and Katcoff, D. J.(2002), ‘Genetic Kinship and Social Structure in a Herd of Square-Lipped Rhinoceroses (Ceratotherium simum simum) at the Zoological Center’, Tel Aviv/Ramat-Gan, Israel Zoo Biology 21:561–571

Shrader, A.M. and Perrin, M.R. (2006) ‘Influence of density on the seasonal utilization of broad grassland types by white rhinoceroses’. African Zoology. 41.2, pp 312-315

Shrader, A.M., Owen-Smith, R.N. and Ogutu, J.O. (2006) ‘How a mega-grazer copes with the dry season: food and nutrient intake rates by white rhinoceros in the wild’. Functional Ecology. 20, pp 376-384

Skinner, J.D., Dott, H.M., Matthee, A. and Hunt, L. (2006) ‘Captive breeding of the white Rhinoceros (*Ceratotherium simum*) and the Cape buffalo (*Syncerus caffer*)’ Onderstepoort Journal of Veterinary Research, 73:237-239

Steuer, P., Clauss, M., Südekum, K.H., Hatt, J.M., Silinski S., Klomburg S., Zimmermann, W., Fickel, J., Streich, W.J. and Hummel, J. (2010) ‘Comparative investigations on digestion in grazing (*Ceratotherium simum*) and browsing (*Diceros bicornis*) rhinoceroses’. Comparative Biochemistry and Physiology - Part A: Molecular and Integrative Physiology. 2010, pp 1-9

Swaisgood, R.R., (2007) ‘Scientific detective work in practice: solving the mystery of white rhinoceros reproduction’. Zoonooz. 80, pp 8-9

Valverde, A., Crawshaw, G.J., Cribb, N., Bellei, M., Gianoti, G., Arroyo, L., Koenig, J., Kummrow, M. and Costa, M.C. (2010) ‘Anesthetic management of a white rhinoceros (*Ceratotherium simum*) undergoing an emergency exploratory celiotomy for colic’. Veterinary Anaesthesia and Analgesia. 37, pp 280-285

Versteege, L. (2010a) ‘*Import’*, De Harpij, 29-1, pp 22-25

Waldram, M.S., Bond, W.J. and Stock, W.D. (2008) ‘Ecological Engineering by a mega-grazer: White rhino, impacts on a South African savannah’. Ecosystems. 11, pp 101–112

4.3 Electronic sources

Anguita – Geerts. P, 24th November 2009. (accessed: 9th April 2010)
Available from: http://tilburgers.nl/?p=4958

EAZA (2018), EAZA Historical Background (accessed: 26 July 2018)
Available from: https://www.eaza.net/about-us/

EAZA (2018a), Taxon Advisory Groups (TAGs) (accessed: 9th of April 2010)
Available from: https://www.eaza.net/conservation/programmes/

IRKA (2010a), Housing (accessed: 28th of June 2010)
Available from: http://www.rhinokeeperassociation.org/facts/white.htm

IRKA (2010b), Training (accessed: 28th of June 2010)
Available from: http://www.rhinokeeperassociation.org/mgmt/training.htm

IRKA (2010c), enrichment (accessed: 28th June 2010)
Available from: http://www.rhinokeeperassociation.org/mgmt/enrichment.htm

IUCN (2010a), Red list (accessed: 16th April 2010)
Available from: http://www.iucnredlist.org/documents/summarystatistics/2010_1RL_Stats_Table_1.pdf

IUCN (2010b), Red list (accessed: 5th April 2010)
Available from: http://www.iucnredlist.org/apps/redlist/details/4185/0

IUCN (2018), Red list (accessed: 6th July 2018)
Available from: http://www.iucnredlist.org/apps/redlist/details/4185/0

RRC (2018) Rhino Resource Centre (accessed: May 2018)
Available from: http://www.rhinoresourcecenter.com/

Species 360 (2018), Species holdings (accessed: 26th July 2018)
Available from: https://www.species360.org/

Tomasova, K., (2006) White rhinoceros, BIAZA, (accessed: June 2010)
Available from: http://www.biaza.org.uk/public/images/campaigns/rhinoDocs/whiteRhino.pdf

WAZA (2018), About WAZA (accessed: 26th Julyil 2010)
Available from: http://www.waza.org/en/site/about-waza

4.4 Unpublished Works

Giesen, M. and Heake, J. (2008) ‘Concept husbandry guidelines for the snow leopard’. European Association of Zoos and Aquaria (EAZA). Amsterdam, Netherlands

Goot, A.C. van der (2009) ‘An analysis on cyclicity and pregnancy in the southern white rhinoceros (*ceratotherium simum simum*) by noninvasive progesterone monitoring using vhf radio telemetry’. Master Thesis for the University of Utrecht and the Institute for Breeding Rare and Endangered African Mammals (IBREAM), pp. 1-14

Griede, T. teacher wildlife management, University of applied sciences Van Hall Larenstein, 13 April 2010

Grün, V. (2006) ‘The influence of faecal scent marks on the behaviour of the white rhinoceros (*Ceratotherium simum simum*)’. Msc thesis, University of Canterbury, Christchurch, New Zealand. pp 1-116

Joseph, S. and Sevenich, M. (1999) ‘Rhino training and enrichment at Disney's Animal Kingdom’ pp 111-120

Lindeman H. 1982. ‘African rhinoceroses in captivity. PhD thesis, University of Copenhagen, Copenhagen.

Nordstrom, L.A.,and Bissonette, J. A. (2006) “Tapirs and rhinoceroses in captivity: an examination of the North American captive populations and their husbandry’. PhD thesis, Utah State University, pp 1-116

Pedersen, G. (2009) ‘Habitat use and diet selection of reintroduced white rhinoceros (Ceratotherium simum) in Pafuri, Kruger National Park’. Msc thesis, Stellenbosch University. pp 1-120

Schelvis, T. and Spijkman, E. (2008) ‘Concept husbandry guidelines for hyenas’ European Association of Zoos and Aquaria (EAZA). Amsterdam, Netherlands

Shrader, A.M. (2003) ‘Use of food and space by white rhinos’. PhD thesis, University of the Witwatersrand, Johannesburg, for the degree of Doctor of Philosophy. Pp 1-190

Versteege, L. Curator Beekse Bergen and co-ordinator white rhinoceros EEP, 7th June 2010b

4.5 Pictures

Title pages	Picture taken by Ripzaad, A. (2009, 2010)
Fig. 1	Boomsma, W. T., and Sijde, van der, M.D. (2010)
Fig. 2	Available from: http://www.desertusa.com/animals/images/white_rhino.jpg (accessed: 30th September 2010)
Fig. 3	Frank Leslie, *The Kingdom of Nature, an Illustrated Museum of the Animal World* (Chicage: Thompson & Thomas, 1900) 182 Clipart ETC is copyright © 2010 by the University of South Florida. Available from: http://etc.usf.edu/clipart/23700/23772/rhino_skelet_23772.htm (accessed: 30th September 2010)
Fig. 4	Emslie, R. and Brooks, M.1999, *African Rhino Status Survey and Conservation Action Plan*, IUCN/SSC African Rhino Specialist group. IUCN , Gland, Switzerland and Cambridge, UK.
Fig. 5 a	Available from: http://lh6.ggpht.com/_bx5BVVH-vKs/SavUgzgXqSI/ AAAAAAAAF-o/L75Sv9JvvUg/PICT0248.JPG (accessed: 7th October 2010)
Fig. 5 b	Available from: http://lh3.ggpht.com/_tlvYZPTcjig/RlYf1O64xmI/ AAAAAAAAADc/Gj8Wt3daFSI/Rhino.jpg (accessed: 7th October 2010)
Fig. 5 c	Picture taken by Andy Laing on 24th of febuari 2009, Blair drummon safari park
Fig. 5 d	Available from: http://laelaps.files.wordpress.com/2007/07/ whiterhino2.jpg?w=510 (accessed: 7th October 2010)
Fig. 5 e	http://farm3.static.flickr.com/2083/2190360724_9746a1c3e1.jpg?v=0
Fig. 6	Available from: http://farm3.static.flickr.com/2502/3730183020_0641bb0564.jpg (accessed: 7th October 2010)
Fig. 7	Boomsma, W.T. (2010)
Fig. 8	Versteege, L. (2010)
Fig. 9	Versteege, L. (2010)
Fig 10	Picture taken by Rotterdam Zoo
Fig. 11	Versteege, L. (2013), in Burgers' Zoo, Arnhem
Fig. 12	Versteege, L. (2013), in Burgers' Zoo, Arnhem
Fig. 13	Versteege, L. (2017)
Fig. 14	Picture taken by Andy Laing on 24th of February 2009, Blair Drummond safari park
Fig. 15	Versteege, L. (2013), in Burgers' Zoo, Arnhem
Fig. 16	Versteege, L. (2013), in Burgers' Zoo, Arnhem
Fig. 17	Picture taken by Jeff Poklen Available from: http://www.biodiversityexplorer.org/mammals/perissodactyla/ images/75623425.KwGJF7Rp_663w.jpg (accessed: 30th September 2010)
Fig. 18	Stevens, C.E. and Hume, I.D. 1995, *Comparative physiology of the vertebrate digestive system*. Cambridge University Press, Cambridge.
Fig. 19	Available from: http://images.travelpod.com/users/milnefamily/1.1248129752. mixed-enclosure.jpg (accessed: 7th October 2010)
Fig. 20	Picture taken by Bela Szandelszky Available from: http://www.sciencedaily.com/releases/2007/10/ 071011212957.htm (accessed: 5th October 2010)
Fig. 21	Picture taken by Pool/Getty Images on 23rd January 2007 Available from: http://cache1.asset-cache.net/xc/73093328.jpg?v=1&c= IWSAsset&k=2&d=77BFBA49EF878921F7C3FC3F69D929FDEA0F15B6548F6517 6965D7A0A881CE7EE26B8B4892D001B0B01E70F2B3269972 (accessed: 7th October 2010)
Fig. 22	Picture taken by Bernd Thissen - European Pressphoto Agency on 21 September 2009 Available from: http://www.worldzootoday.com/2009/09/21/your-morning-adorable-keepers-bottle-feed-rhinoceros-calf-in-german-zoo/

(accessed: 12th September 2010)

Fig. 23 Available from: http://news.bbc.co.uk/nol/shared/spl/hi/pop_ups/08/uk_enl_1251467279/img/1.jpg (accessed: 6th October 2010)

Fig. 24 Hadley, K. (2000) 'Scent Preferences of Southern white rhinos'. Shape of enrichment. 9.4, pp 1-3

Fig. 25 Available from: http://img2.photographersdirect.com/img/12366/wm/pd_304308.jpg (accessed: 7th October 2010)

Fig. 26 IATA guidelines for transport, pp 253

Fig. 27 Picture taken by Emma Tannebaum
Available from: http://media.al.com/birmingham-news/photo/a91f3c148ce4fb7bd255316309db6ea1.jpg (accessed: 12th September 2010)

Appendix I: EAZA standards

EAZA standards for the Accommodation and Care of Animals in Zoos and Aquaria

Approved by EAZA Council on 27 September 2014 -

Introduction

These standards are based on present knowledge and practice for the accommodation and care of animals in zoos and aquaria.

In this Annex the following definitions shall apply:

1. **Zoos and aquaria** refer to all establishments open to and administered for the public to promote nature conservation and to provide education, information and recreation through the presentation and conservation of wildlife. This definition shall include zoos, animal parks, safari parks, bird gardens, dolphinaria, aquaria and specialist collections such as butterfly houses as defined in article 2 of Council Directive 1999/22/EC of 29 March 1999;

2. **Zoos and aquaria** situated in EU countries are requested to have a valid license under Council Directive 1999/22/EC of 29 March 1999. All others need valid licenses to operate, if these exist. The dates and/or numbers of these licenses have to be registered with the EAZA Executive Office;

3. **Animals** shall refer to all species of the animal kingdom including species of the classes mammals, birds, reptiles, fish, amphibians and invertebrates;

4. **Welfare** shall refer to the physical, behavioural and social well-being of animals through the provision of appropriate conditions for the species involved, including but not necessarily limited to housing, environment, diet, medical care and social contact where applicable;

5. **Enclosure** means any accommodation provided for animals in zoos and aquaria;

6. **Enclosure barrier** means a barrier to contain an animal within an enclosure;

7. **Stand-off barrier** means a physical barrier set back from the outer edge of an enclosure barrier designed to prevent public access to the latter;

8. **Hazardous animals** means any representative of the groups or species listed in Annex 1 and any other animal which, because of its individual disposition, sexual cycle, maternal instincts, or for any other reason, whether by biting, scratching, butting, compression, injecting venom or by any other method, is likely to injure seriously or transmit disease to humans;

8. **Dangerous carnivores** means all members of the genera Panthera, Acinonyx, Lynx and Neofelis, the families Ursidae and Hyaenidae, Canis lupus, Canis rufus and Lycaon pictus.

1. Animal care – Welfare, health and hygiene

1.1 Routine observation of the animals

1. The condition and health of all animals in the zoo to be checked daily by the persons in charge of their care for that particular day.
2. Any animals which are noted to be unduly stressed, sick or injured to receive immediate attention and, where necessary, treatment.

1.2 Accommodation - Space, Exercise and Grouping

EAZA members are expected to provide a high standard of accommodation for all the animals in their care, both on-show and off-show, permanent and temporary. EAZA Best Practice Guidelines (when available) should be consulted when available and their recommendations implemented wherever possible. Accommodation must take account of the welfare of the species, their space and social needs, appropriate management by staff and appropriate display to the public. Important considerations that must be taken account of are:

1. Provision of a physical space that is appropriate to the species, taking account of their three-dimensional needs.
2. Animals to be provided with an environment, space and furniture sufficient to allow such exercise as is needed for the welfare of the particular species.
3. Design must take account of the management needs of the species. This includes:
   a. Safe and appropriate presentation of food and water.
   b. Management of social conflict through separation areas, visual barriers and other means, including accommodation for animals temporarily separated from a group.
   c. Catch-up facilities.
   d. Facilities for the management of breeding animals, such as cubbing dens, nest sites or spawning substrates.
   e. Introduction, quarantine and health-care facilities.
   f. A safe and appropriate cleaning regime.
   g. Environmental and behavioural enrichment.
4. Enclosures to be of sufficient size and animals to be so managed -
   a. to avoid animals within herds or groups being unduly dominated by individuals;
   b. to avoid the risk of persistent and unresolved conflict between herd or group members or between different species in mixed exhibits
   c. to ensure that the physical carrying capacity of the enclosure is not over-burdened;
   d. to prevent an unacceptable build-up of parasites and other pathogens.
5. Animals should never be unnaturally provoked for the benefit of the viewing public.
6. Animals in visibly adjoining enclosures to be those which do not interact in an excessively stressful way.
7. Provision of facilities for good observation of the enclosure and its animals by staff and researchers.
8. Provision of a high standard of public viewing experience, that demonstrates fully the animals and their behaviours, and which is consistent with the educational messages and strategies relevant to the species, the organisation and EAZA.

1.3 Accommodation - Comfort and Well-being

1. The environmental conditions, including temperature, humidity, ventilation, seasonal changes and lighting of enclosures to be suitable for the comfort and well-being of the particular species of animal at all times, including holding quarters and/or off-exhibit housing and in particular -
   a. consideration to be given to the special needs of pregnant and newly-born animals;
   b. newly-arrived imported animals to be fully acclimatized bearing in mind that this may be

only a gradual process;

c. tanks for fish and aquatic invertebrates to be adequately oxygenated, and appropriate water quality to be provided.

d. Animals in outdoor enclosures to be provided with sufficient shelter from inclement weather or excessive sunlight where this is necessary for their comfort and well-being.

1.4 Social group management

1. Facilities for keeping animals shall allow maintaining a social unit that reflects the life history of a given species in the wild, and thus may need to have sufficient flexibility to adapt towards changing group dynamics.
2. Before introducing a new individual to a social group, an assessment should be made of its adaptability to the group and consequently the implications for its individual welfare, as well as the welfare of the animals in the social group.

1.5 Encouragement of natural behaviour and minimising of unnatural behaviour

1. Animals kept in EAZA collections should be encouraged to perform as much of their natural behavioural repertoire as possible and acceptable. Whenever possible unnatural behaviour should be prevented or actively discouraged. Important elements in achieving this are enclosure design, environmental and behavioural enrichment and feeding regimes (see above).

1.6 Furnishings within Enclosures

1. Design must take account of behavioural needs and behavioural management of the species, allowing adequate spatial separation between individuals or subgroups.
2. Provision of a rich environment of appropriate structures within the space that enable the animals to express their behavioural repertoire as fully as possible.
3. Animal enclosures to be furnished, in accordance with the needs of the species in question, with such items as bedding material, perching, vegetation, burrows, variety of substrates, climbing structures, appropriately designed nest boxes, refuges and hiding places and pools.
4. Provide appropriate environmental and behavioural enrichment.

1.7 Mixed exhibits

1. Housing animals in mixed species exhibits can create a more natural display of animals for the visitors, potentially enrich the lives of species or individuals in the exhibit and provide space necessary for population management.
2. When exhibiting animals in a mixed species enclosure EAZA members should ensure that housing conditions are an acceptable standard for the welfare of all species exhibited. The potential positive and negative welfare implications for species combinations should be carefully assessed prior to introduction and monitored after the introduction. At the very least the following points must be part of the assessments:
3. Trauma risk;
4. Hierarchy and potential nutrition-related problems;
5. Transmissible viral and bacterial diseases;
6. Parasitic disease transfer between species.
7. EAZA members shall conduct research into the successes and failures of mixed species exhibits and share information within the community.

1.8 Free ranging species

1. If animals are being kept free ranging in a zoo, the potential for containing them safely should always be available and the animals should be used to entering this enclosure for management purposes (e.g. veterinary examinations and collection management).
2. Escape into the wild and/or crossbreeding with local species must be avoided at all times.

1.9 Hand rearing

1. Natural rearing is preferred over hand rearing whenever possible. EAZA members should minimise the need for hand rearing by providing appropriate accommodation and care for the individual animals and when applicable the social group.
2. When natural rearing cannot be achieved EAZA members shall assess the need and possibilities for hand rearing considering potential behavioural implications for the individual, the importance for the social group and the overall population.
3. Euthanasia may be an alternative to hand rearing particularly when introduction potential into the group is low, behavioural problems are expected and/or when sufficient future ”quality of life” cannot be guaranteed.
4. In certain species foster rearing could also be considered as an alternative to hand rearing. If EEP species are concerned the coordinator should be consulted before making any decision (time allowing).
5. If hand rearing is decided upon it should be aimed at raising the individual(s) such that it develops as much species-specific behaviour as possible and the animal(s) should be introduced back to its conspecifics as soon as possible.
6. EAZA members should focus any communication related to hand rearing on the biological considerations and purpose of hand rearing the animal, and avoid anthropomorphic interpretations for the purpose of commercial gain.

1.10 Prevention of Stress or Harm to Animals

1. Enclosures and barriers to enclosures to be maintained in a condition which presents no likelihood of harm to animals, and in particular -
   a. any defect noted in an animal barrier or in any appliances or equipment within animal enclosures to be repaired or replaced without delay;
   b. any defect likely to cause harm to animals to be rectified at once or, if this is not possible, the animals to be removed from the possibility of any contact with the source of the danger;
   c. any vegetation capable of harming animals to be kept out of reach.
2. All plant and fixed equipment, including electrical apparatus, to be installed in such a way that it does not present a hazard to animals and its safe operation cannot be disrupted by them.
3. Rubbish in animal enclosures to be cleared regularly to avoid any possibility of harm to animals.
4. Trees within or near animal enclosures to be regularly inspected and lopped or felled as appropriate to reduce the risk of damage to enclosure barriers and animals being harmed by falling branches or using trees as a means to escape.
5. Smoking is prohibited in animal enclosures, in parts of buildings where animal enclosures are located and in areas where food is stored or prepared.
6. Animals to be handled only by, or under the supervision of, competent trained authorised staff; and this to be done with care, in a way which will avoid unnecessary discomfort, behavioural stress or actual physical harm to animals.
7. Any direct physical contact between animals and the visiting public only to be under the control of zoo staff and for periods of time and under conditions consistent with the animal’s welfare and not leading to their discomfort.
8. When organising events, EAZA members must ensure these are organised and performed such that they keep the disturbance to the animals at a minimum. The effect of events on the welfare of individual animals should be assessed and changes implemented where necessary.

1.11 Training

Training falls broadly into two categories namely for ‘Husbandry Purposes’ and for ‘Public Animal Handling Presentations & Shows’

1.11.1 Animal training for husbandry purposes
Training as part of some husbandry routines can be helpful in reducing stress and to facilitate management. Physical or chemical restraint of zoo animals can be stressful and the necessity for it can often be avoided by training animals to cooperate with certain husbandry routines.
EAZA supports the training of animals for husbandry purposes provided that:

1. Only positive reinforcement techniques are used (i.e. rewards and never punishments) and the animals are never forced to take part in training.
2. The training goals are not detrimental to the welfare of the animal or its conspecifics.
3. The training process is regularly monitored and reviewed to ensure that the safety of staff and the welfare of the animals are maintained.

1.11.2 Animal training for public demonstrations and ambassador animal interactions
Animal displays for the public can be a valuable part of the zoo visit experience in that they allow visitors to get close to, and even have contact with, animals, providing a powerful tool to help inspire them to care about and protect wildlife.
EAZA supports the use of animals in animal presentations and shows provided that:

1. The content of the display is educational and designed to demonstrate, and inspire respect for, the animals' natural behaviour and abilities.
2. The content of the display does not demean or degrade the animal and is not detrimental to its welfare.
3. Training animals for the display is carried out according to the same basic welfare principles as those for husbandry purposes (including point 1a-c above).
4. Animals are not specifically separated from their dams for hand rearing for the sole purpose of producing tame individuals for use in public demonstrations or guest interactions.
5. When not participating in shows the animals are housed appropriately and meet (wherever possible) EAZA Best Practice Guidelines for the species (e.g. environmental enrichment is provided even if they are being held in more functional accommodation).
6. In the case of external contractors, the source of the animals and the conditions in which they are kept should meet the same high standards as those required by the EAZA member zoo for their own stock.
7. The animals are given appropriate numbers of rest periods between handling sessions and are not used during times at which they may be more susceptible to stress such as pregnancy, moulting, sloughing etc.
8. When animals are used in presentations or shows this should not conflict with the objectives and requirements of EEP/ESB programmes. The relevant coordinator/studbook keeper should be consulted before any decision is made on introducing an individual programme animal into a show.

1.12 Food and Drink

1. A diet is considered to be "nutritionally balanced" when it provides appropriate levels of known dietary essential nutrients based on current knowledge and information.
2. Veterinary or other specialist advice to be obtained and followed concerning all aspects of nutrition.
3. Food and drink provided for animals to be of the nutritive value and quantity required for the particular species and for individual animals within each species, bearing in mind the condition, size and age of each animal; the need to allow for special circumstances (e.g. fast days or longer periods of fast or hibernation) and special diets for certain animals (e.g. animals undergoing a course of veterinary treatment, or pregnant animals).
4. A nutritionally balanced diet must be provided in a suitable form and correct proportion based on the most appropriate behavioural and physiological needs of the species.

5. A nutritionally complete diet should also stimulate natural feeding behaviours encouraging, whenever reasonably possible, the animals to obtain food in a manner similar to that in the wild and should allow a similar amount of time to be spent on feeding.
6. Food used for animals should be of adequate quality and checked before it is used to ensure it is in good condition and appropriate to feed.
7. Zoos and aquaria should put effort into ensuring foods have a sustainable origin, wherever possible.
8. When feeding live prey, local legislation must be followed, and the welfare of the prey must be taken into consideration to ensure stress is minimised.
9. Uncontrolled feeding by visitors is not permitted. Where public feeding is allowed this should be supervised and the fed items must be quantified as part of the diet for the animals.
10. Supplies of food and drink to be stored, prepared and offered to the animals under hygienic conditions.
11. Natural behaviour of the animals, particularly social aspects to be considered when offering food and drink, and feeding and drinking receptacles if used, to be placed so as to be accessible to every animal kept within a particular enclosure.

1.13 Sanitation and control of disease

Proper standards of hygiene, both in respect of the personal hygiene of the staff and that of the animal enclosures and treatment rooms, to be maintained, and in particular -

1. Special attention to be given to the cleaning of animal enclosures and equipment within them, to reduce the risk of disease or disease transfer, including in the case of aquatic animals, regular monitoring of water quality;
2. Non-toxic cleaning agents to be readily available, along with supplies of water and the means to apply them
3. Veterinary advice to be obtained and followed regarding all cleaning and sanitation requirements of enclosures or other areas following identification of an infectious disease in any animal.
4. The drainage of all enclosures to be capable of removing efficiently all excess water.
5. Any open drains, other than those carrying potable water, to be outside the areas to which animals have access.
6. Refuse material to be regularly removed and disposed of.
7. A safe and effective programme for the control of pests and, where necessary, predators to be established and maintained throughout the institution. Animals must not escape from the zoo or aquarium and create an ecological threat for native wild species.
8. Keeper staff to be instructed to report immediately if they have contracted or are in contact with any infection which they have reason to believe could be transmitted to, and adversely affect the health of, any animal; and management then to take appropriate action.
9. Keeper staff to be instructed to report in confidence any other disability which might affect their capacity to manage the animals in a safe and competent manner; and management then to take appropriate action.

2. Animal Care – Veterinary Aspects

2.1 General Veterinary care

1. Arrangements to be made for routine veterinary attendance. In case of fishes and invertebrates, other specialist attendance is also acceptable. This also applies to all other references to veterinary aspects in fishes and invertebrates in this document.
2. A programme of veterinary care to be established and maintained under the supervision of a veterinary surgeon or practitioner.

3. Routine examinations, including parasite checks, to be carried out and preventive medicine, including vaccination, to be administered at such intervals as may be recommended by a veterinary surgeon or practitioner.
4. Where a full veterinary service is located at the institution, the facilities to include: an examination table; a range of basic surgical instruments; anaesthetic facilities; basic diagnostic instruments; sufficient power points to take light and other electrical fittings; facilities, where appropriate, to take blood and other samples and to prepare and dispatch them; and a comprehensive range of drugs.
5. Where a full veterinary service is not available at the institution, a treatment room to be provided at the premises for use where appropriate for the undertaking of routine examination of animals in clean, ventilated surroundings.
6. A room or rooms to be provided for the care of unduly distressed, sick and injured animals.
7. Facilities to be available for collecting, restraining and, if necessary, for administering a general anaesthetic, for euthanizing animals and for the after-care of animals recovering from sedation.
8. Reserve accommodation to be available, away from other animals, for the isolation and examination of newly-arrived animals, under quarantine restrictions (conditions) where necessary.
9. Newly-arrived animals to be kept isolated as long as is necessary to ensure proper examination before introduction to other animals in the collection.
10. Particular attention to be paid to hygiene in the quarters where isolated or quarantined animals are kept.
11. Where practicable, protective clothing and utensils used by staff in the isolation area should be used, cleaned and stored only in that area.
12. All animal drugs, vaccines and other restricted veterinary products to be kept safely under lock and key with access by authorised persons only.
13. Except under the direction of a veterinary surgeon or practitioner, members of the staff of the zoo not to possess or administer controlled drugs.
14. Zoo management to seek agreement with the consulting local veterinary adviser regarding the desirability of either the zoo or aquarium, a local hospital or the veterinary surgeon or practitioner himself, of holding supplies of antidotes to potentially toxic veterinary products used at the institution.
15. All unwanted, contaminated veterinary equipment to be disposed of safely and following relevant legislative prescriptions.

2.2 Mutilation

1. Mutilation of any animal for cosmetic purposes, or to change the physical appearance of the animal, is not acceptable. There should be a net welfare benefit to the individual animal and/or its conspecifics before accepting mutilation for educational or management reasons. This also includes pinioning of birds. Closed aviaries of appropriate size are thus preferred to open enclosures where pinioning is the only efficient method of restraint.
2. Marking of animals for identification reasons should always be carried out with the least harmful method available and should only involve mutilation of any sort when no other method has proven feasible. Where mutilation is used it must always be carried out in accordance with approved veterinary protocols.

2.3 Post-Mortem Facilities

1. Dead animals to be handled in a way which avoids the risk of any transmission of infection.
2. The cause of death for each animal dying in the collection needs to be established where reasonable and practicable to do so, including, in the majority of cases, the examination of the specimen by a veterinary surgeon, pathologist or practitioner with relevant experience and training.

3. Institutions should communicate causes of death of programme animals to the EEP coordinator or studbook keeper.
4. Where animal carcasses cannot be quickly removed to a professional veterinary laboratory centre outside the premises, facilities should be provided for conducting post-mortem examinations and the processing of samples resulting from them in a safe and hygienic manner. If immediate post-mortem examination is not possible, then in consultation with the veterinary surgeon or practitioner, refrigerated facilities or a deep freeze for storage to be provided pending the removal in a suitable insulated container to a post-mortem laboratory.
5. Facilities and equipment in any room provided on the premises for post-mortem examinations to include: an efficient drainage system; washable floors and walls; an examination table; an adequate selection of appropriate instruments; facilities for taking and preserving specimens; and, if larger animals are kept in the collection, a hoist.
6. Following post-mortem examinations conducted on the zoo premises, carcasses and organs to be removed swiftly and disposed of safely.

3. Population Management

3.1 Reproduction

1. Reproduction is an integral part of the quality of life and natural behaviour of each living animal. However, offspring of zoo animals should not be produced if they cannot be kept in house or placed in other suitable conditions.
   The following measures can be considered to limit undesired reproduction:
   a. temporary separation, or contrarily in some cases the continued holding together, of males and females
   b. holding of only one sex of a species; all-male or all-female groups
   c. extension of birth intervals
   d. removal, shaking or freezing (of part) of egg clutches
   e. hormone injections, oral medication, or implants in females

f. PZP vaccinations of females
g. sterilization of males/females, including vasectomy and castration.

3.2 Transfer and disposition of animals

1. Members should ensure that institutions receiving animals have appropriate facilities to hold the animals and skilled staff who are capable of maintaining the same high standard of husbandry and welfare as required of EAZA members (in this context circuses would not be regarded as appropriate recipients of animals from EAZA members).
2. Any details of health, diet/nutrition, reproductive and genetic status and behaviour that might affect management of an animal being transferred (or other animals in the group at the receiving institution) should be disclosed at the commencement of negotiations.
3. All animal transfers should conform to the international standards applying to the particular species. Where appropriate, animals should be accompanied by qualified staff and/or timely information provided that will facilitate the animal's adjustment to its new home.
4. For the benefit of the future viability of EAZA/EEP populations, all transfers of EEP animals must be arranged in full consultation with, and the agreement of, the EEP Coordinator.
5. In order to ensure the non-commercial status of EEPs any selling of EEP animals must be avoided.

3.3 Euthanasia

Euthanasia as a structural solution for undesired surplus animals may be acceptable under certain conditions beyond veterinary indication, such as the following:

1. Animals that can/may no longer make a breeding contribution, for example because of old

age, genetic over-representation of the possession of undesirable inheritable genetic traits.
2. Young animals born despite reproduction-limiting measures or recommendations that have been recently born, have reached weaning age or another age in which they would naturally leave the parent(s) or natal group.
3. Incompatibility of an animal with its conspecifics.
4. Hybrids and animals of an unknown or undefined subspecies in cases in which this is considered of importance.
5. Animals that are more dangerous than is reasonably expected.
6. Animals that despite changes in conditions (e.g. institution/enclosure/diet) continue to display abnormal behaviour or extraordinary timidity.
7. Animals that for some reason cannot otherwise be placed in suitable facilities.
8. Donated or otherwise acquired injured rehabilitation animals.

3. Safety and Security

4.1 General provisions

1. Local safety and security legislation regarding zoos and aquaria must be applied.

4.2 Enclosures

1. Other than when elsewhere in the control of authorised staff, animals kept for exhibition in the zoo to be kept at all times in enclosures or, in the case of free-running non-hazardous animals, within the perimeter of the zoo.

4.3 Enclosure barriers

1. Enclosure barriers to be designed constructed and maintained to contain animals within the desired enclosures.

4.4 Stand-off Barriers

1. Where direct contact would be possible between visitors and hazardous animals through or over any enclosure barrier, to the extent that such an animal would be capable of causing injury, a stand-off barrier to be provided sufficiently far back to prevent such contact.

4.5 Perimeter Boundaries

1. The perimeter boundary, including access points, to be designed, constructed and maintained to discourage unauthorised entry and, so far as is reasonably practicable, as an aid to the confinement of all the animals within the perimeter of the institution.
2. No perimeter barrier to include any electrical section less than 2 metres from the ground, except in those cases where it also serves as a normal animal barrier and cannot be reached by the visiting public.

4.6 Warning Signs

1. In addition to a stand-off barrier, an adequate number of clearly visible safety signs to be displayed at each enclosure where there may be significant danger, including electric fences.

4.7 Exits

1. Sufficient exits from the zoo or aquarium to be provided, having regard to the size of the institution and the number of visitors anticipated at any time that may need to leave quickly in an emergency.
2. Exits to be clearly signposted and marked.
3. Each exit from the zoo or aquarium to be kept clear and to be capable of being easily opened

from inside to allow the release of persons from the institution. All such gates to be capable of being closed and secured to discourage the escape of animals.

4.8 Drive-Through Enclosures

1. Unless there is stricter local legislation, this chapter will be applied to drive-through enclosures.
2. Where dangerous carnivores are kept in drive-through enclosures, entry and exit to such enclosures to be through a system of double gates, with sufficient space between to allow the gates to be securely closed to the front and rear of any vehicle which may enter or need to enter the enclosures.
3. In the case of dangerous carnivores the access gates to be protected by fencing positioned at right angles to the perimeter fence on each side of the roadway with the enclosure, and of the same standard as that for the main enclosure barrier and extending back from the access for a distance of at least 25 metres.
4. Double gates to be designed and maintained so that, where hazardous animals are within or have access to the enclosure secured by the gates, one gate cannot be opened until the other has securely closed - though, provided no danger to the public is thereby caused, provision may be made for this arrangement to be overridden in the event of an emergency arising.
5. For other hazardous animals, except those grazing or hoofed animals where a cattle grid would be sufficient to contain them, single entry/exit gates, supervised at all times, to be provided.
6. Access points between enclosures to be controlled to prevent animals entering adjoining enclosures.
7. Electrified pressure pads, where used, to be designed and installed to ensure that in the event of their failure, any gate they control will close automatically or otherwise operate to ensure that animals are safely secured within their enclosure.
8. Gates which are mechanically-operated to have an alternative method of control whereby they can be opened and closed manually in the event of an interruption of the power supply or other emergency and to be designed to close automatically when subject to power failure.
9. Operators of mechanically-operated gates to have a clear, unobstructed view of the gates under their control and of the area within the vicinity of those gates.
10. A one-way road system to be used to assist the traffic flow and thus reduce the risk of accidents.
11. Stopping to be permitted only at places where the road is at least 6 metres wide.
12. Where dangerous carnivores and primates and (except where the enclosure is supervised by competent staff in a manner which prevents any danger to the public) any other hazardous wild animal are kept:
    a. no vehicle to be allowed access unless a rescue vehicle capable of effecting its recovery is immediately available;
    b. access to vehicles without a solid roof to be prohibited at all times;
    c. notices, which are readily visible and easy to read, to be displayed to warn visitors whilst in the enclosure to:
        i) Stay in vehicle at all times;
        ii) Keep all vehicle doors locked;
        iii) Keep vehicle windows and sun-roof closed;
        iv) Sound the horn or flash the headlights and await the arrival of a rescue vehicle if they break down.
13. Continuous observation to be maintained over the entire area of each enclosure containing any hazardous animal.
14. The staff member in overall control of supervision to be armed with an appropriate firearm and to be trained in its use so that a hazardous animal can be killed in an emergency if this will save human life or injury.

4.9 Removal of animals from enclosures

1. Hazardous animals not to be allowed out of their usual enclosures for the purpose of direct contact with the public, except, where the zoo operator is satisfied that such animals are not, when under control, likely to cause injury or transmit disease.
2. Where hazardous animals are allowed out of their usual enclosures an authorised and experienced member of the staff to accompany each animal.
3. Zoo operators to exercise caution and discretion in the case of the removal of non-hazardous animals since the behaviour of all animals may be less predictable when away from their usual enclosures.
4. Precautions to be taken to avoid injury to visitors when animals are used for rides.

4.10 Escape of animals from their enclosures

1. Zoo operators to assess whether any danger may arise in the event of an animal escaping from its enclosure and to consider the possible or likely attempted escape route within and from the institution if this were to happen.
2. In the case of the escape of animals emergency plans must be available and fully understood and practised by all staff.
3. This emergency plan should include a member of staff to be readily available at all times to take decisions regarding escaped animals, including the use of firearms if needed.
4. Every employee with tasks under the emergency procedures to undergo periodic refresher training and practice.

4.11 Safety of access for the public

1. Buildings, structures and areas to which the public has access to be maintained in safe condition.
2. Trees within areas where visitors are likely to be walking or sitting to be regularly inspected and lopped or felled as appropriate to avoid visitors being harmed by falling branches etc.
3. Warning to be given of all edges where a person might fall, including into water; and, where necessary, such edges to be guarded by a barrier which would be capable of restraining children from falling.
4. Each walkway over an animal enclosure to be designed, constructed and maintained to withstand safely the weight of the maximum of adults who could use it at any time; and maintained, sited or protected so as to withstand any contact by hazardous animals and prevent contact between such animals and visitors.
5. The visiting public not to be allowed to enter any buildings or other areas of the zoo premises which could present an unreasonable risk to their health and safety.
6. Any buildings to which visitors are not allowed on the grounds referred to above, to be kept locked and warning notices to be displayed to indicate that access is both unsafe for, and not permitted to, the public.
7. Other areas to be clearly defined (e.g. by means of barriers and similar warning notices), or by suitable notices together with road markings where frequent access is necessary for vehicles operated by zoo staff along roadways to which the public are not admitted.

4.12 Emergency First-Aid

1. First-aid equipment and written first-aid instructions to be readily accessible on the premises.
2. Where venomous animals are kept, the appropriate and up-to-date anti-venom to be held at the zoo or a local hospital or within a reasonable time frame ensuring the safety of staff and visitors, and kept in accordance with the manufacturer’s instructions.
3. Written instructions to be provided for staff on the procedure to be followed in the event of an incident involving any venomous animal and a visitor or another staff member. These instructions would include immediate care instructions, the telephone number of the

nearest hospital and poisons centre, and normal emergency contacts. A pre-prepared form that can be sent with the patient being sent to the local hospital should include:

a. the nature of the bite or sting and the species inflicting it;
b. the specification, for cross-reference purposes, of the anti-venom which accompanies the patient;
c. the telephone number of the nearest poisons centre;
d. the telephone number of the institution.

5. Miscellaneous

5.1 Insurance against liability for damage or injury caused by animals

1. Zoo operators to hold a current liability insurance policy or other legal arrangements which indemnifies them and every other person under a contract of service or acting on their behalf, against liability for any damage or injury which may be caused by any of the animals, whether inside or outside the zoo, including movement by vehicle. Any upper limit on the sum involved which is included in the terms of such insurance to be set at an adequate and realistic level.

5.2 Stock records

1. Animal records are to be kept on a computer system using the ARKS software, or Zoological Information Management System (ZIMS), and to be included on the global zoo animal database of Species360, by means of which information can be quickly retrieved.
2. Alternatively, records may be kept by means of an established and globally recognised and accepted record system and maintained in relation to all individually recognised animals and groups of animals.
3. Where animals are disposed of or die, the records to be kept in the appropriate recording system as described in Article 95.
4. The records should provide the following information -
   a. the correct identification and scientific name;
   b. the origin (i.e. whether wild or captive born, including identification of parents, where known, and previous location/s, if any);
   c. the dates of entry into, and disposal from, the collection and to whom;
   d. the date, or estimated date, of birth;
   e. the sex of the animals (where known);
   f. any distinctive markings, including tattoo or freeze brands etc.;
   g. clinical data, including details of and dates when drugs, injections, and any other forms of treatment were given, and details of the health of the animal;
   h. the date of death and the result of any post-mortem examination;
   i. the reason, where an escape has taken place, or damage or injury has been caused to, or by, an animal to persons or property, for such escape, damage or injury and a summary of remedial measures taken to prevent recurrence of such incidents.
5. In addition to the individual records, an annual stock list of all animals to be kept preferably in the form given below. (Estimated numbers should be available for all fish and invertebrate species).
   a. Common and scientific names of the species
   b. Total in the collection at 1 January
   c. Number of arrivals into the collection from all sources during the year
   d. Number of births into the collection during the year
   e. Number which died within 30 days of birth
   f. Number which died older than 30 days after birth/hatching
   g. Number departed collection, including sales, breeding loans, etc.
   h. Total remaining in the collection at 31 December

This record, giving details of male/female/unsexed animals as appropriate, to be set out in columns for ease of compilation and reference, e.g.:

Common name	Scientific name	Group 1-1-05	Arrive	Born	Neonatal death	Death	Depart	Group 31-12-05
Bennett's wallaby	Macropus rufogriseus	5.11.0	1.0	1.1.8	1.0.3	1.2.0	1.1	4.9.5

6. All records can be kept in the local language or in the English language (in order to facilitate the international exchange of information and cooperation).
7. Surplus animal stock only to be passed on to responsible facilities which have the appropriate facilities and expertise).

5.3 Transportation and Movement of Live Animals

1. Facilities suitable for hoisting, crating and transportation of all the kinds of animals kept within the zoo, to destinations both inside and outside the zoo, to be available if not kept at the zoo.
2. Any animal taken outside the zoo to be in the personal possession of the operator of the zoo, or of competent persons acting on his behalf, and adequate provision to be made for its safety and well-being at all times.
3. Any hazardous animal taken outside the zoo to be kept securely at all times. Such animal to be kept away from direct contact with persons other than the zoo operator or competent persons acting on his behalf, except where the zoo operator is satisfied that it is not likely, when under control, to cause injury or transmit disease.

Appendix II: Rhinos and training

To improve rhino management, keepers have developed training guidelines to meet a variety of husbandry needs and provide mental stimulation. A well-managed program would enhance the success of most non-invasive veterinary procedures, as well as permit the execution of more intensive practices. In general, the same training principles can be applied to all rhino species, but it is imperative that the keepers are consistent and follows the institution's training protocols.

Description of Selective Behaviours and Their Commands

Target - This command is used for positioning of the rhino. The correct response is to touch the target with its upper lip. The target is placed at the location where the trainer wants the rhino positioned and when the rhino approaches the target it will touch the target with its upper lip.

Lean In or Over - This command is used for lateral positioning the rhinos for blood draws, and can be used to evaluate the condition of the rhino. To get the animal to perform this behaviour the trainer uses the command "target" to line the animal up, and then holds the target to the animal's hip or shoulder. When the "over" command is given, the animal should then side step towards the target. The correct response to this command is to bring the targeted side of the rhino towards the barrier (being lined up parallel with the barrier), with the hip or front shoulder actually touching the target. The animal can be stopped at any point by using the bridge, but is generally asked to step all the way to the barrier so the animal's side is in contact with it.

Back side

Front Shoulder

Open - This command is used for checking the mouth for gum coloration, presence of lesions or sores, or general dentition inspection. With this command, the trainer targets the animal's head into proper position, and then issues the command "open". The correct response for this command is to have the mouth open far enough to check the animal's teeth. Some trainers actually use the physical cue to touching the upper and/or lower lip(s) as the signal with the "open" command. Then use the "hold" command to maintain the mouth in desired position.

Contents

Foot - This command can be used for positioning desired leg for phlebotomy procedure and can be used to perform any necessary footwork. Place the block in front of the desired foot (usually the foot closest to trainer), and give the command “foot” to signal lifting of foot onto the block and placing it flat on the surface. Once the foot is on the block, the trainer then places a hand on top of the foot while saying, “steady”. The rhino should keep the foot on the block until the release command is given. The foot can be shifted forward on the block to facilitate easy filing of the nail(s). Animals can also be conditioned to permit placement of nails on block so their pad can be thoroughly inspected and trimmed, as needed.

Steady - This command is given when the trainer needs the rhino to hold position for a certain amount of time. For example: When drawing blood, steady is used to keep the animal’s leg in position while blood is being drawn.

Phlebotomy procedure (front leg) – Rhinos are generally conditioned to either place legs in desired position or place foot on block for phlebotomy procedures. Initially trainer will need to condition for positioning, then steady command and finally desensitization to venipuncture. Desensitization to venipuncture can start as simple as touching the inner surface of front leg opposite trainer and progress to use of blunted needle, etc. to simulate the pressure and device(s) used for procedure. Remember to bridge and reward animal for maintenance of position during this process since that will be a required behaviour for this process. Once you progress to the actual venipuncture itself, you may have to do initial stick, reward/re-focus animal, repeat steady command, and then finally proceed to repositioning needle for phlebotomy process. In most rhino species proper conditioning will enable blood samples to be taken from the ear, base of tail, front or rear legs.


Vein on opposite leg


Initial Stick

Down - The correct response to this command is to lie down sternal, parallel to the bars. To get some animals to lie down the trainer brings either side of the rhino over to the barriers, using the command “over”, and then use a physical cue such as rubbing the rhino’s back or the inside of the back legs to encourage them to lie down. The final result will be animals placing themselves in a sternal position parallel to barrier. This behaviour has so been trained via shaping and the use of the “target”.

Cue – Rubbing the rhino’s back


Result Sitting Position

Chute/restraint Training

The use of a chute to restrain and/or confine the animal can be helpful in preventing the rhino from moving excessively during a procedure, which could cause injury to the animal or trainer. The first step in restraint training is getting the animal comfortable entering the device. Animal can either be “targeted” into the chute, or use successive approximation to progress to the end goal of entering the chute. Depending on the construction of your particular restraint, it may advantageous to have restraint in the fully open position during this initial phase then progress to shutting the front door, and finally progressing to full confinement. To shape the entrance behaviour, the animal is rewarded for making progress towards the chute. For example, if the animal is standing just outside the chute, for each step it takes towards the chute the animal is bridged and rewarded until it is completely within the restraint.

Once the animal is in the restraint, it is a good idea to get the animal comfortable remaining in the chute for a period of time and “bridging” for calm demeanour while within the restraint before training any other behaviour within the device.

Trainer must remember to desensitize animal (and reward calm demeanour and attitude) to the visual and auditory sounds of door movement in front and behind animal and any other moving parts of the device before progressing to training additional behaviours.

Some common commands and behaviours associated with the chute are:

- **Steady**: Command given to have the animal remain in the desired position (i.e. if the rhino's foot is on a block).
- **Over or Lean in:** Command given to have the animal step, or move the entire body closer to the wall of the chute.
- **Back**: Command given to the have the animal step backwards, either for positioning or exiting the chute.
- **Move Up**: Command given to have the rhino step forward for positioning, can also be used to enter/exit the chute.
- **Foot**: This is used generally to place a foot on a block, or to position it in a way to draw blood.
- **Ear**: Command given to relax the ear to draw blood.
- **Tail**: Command given to relax the tail, which allows the trainer the ability to manipulate the tail in the desired position, either for blood draws, or rectal ultrasound/palpations.

(IRKA, 2010b)

Appendix III: EAZA Bio Bank Sampling protocol

Sampling protocol

Live animals

Samples should be taken in accordance with national legislation.

Whole blood (max 5 ml), in plastic EDTA or PAXgene blood collection tubes. Invert 15 times to mix.

Or

Tissue (max 1cubic centimeter) from e.g. skin, muscle, or umbilical cord. Placed in a plastic tube (2ml screw cap) containing 70% ethanol or frozen in a plastic bag.
Do not use formalin or methylated alcohol.

Serum (1-10 ml) in plastic tubes. Must be spun and seperated. Should only be provided if it is accompanied by a blood or tissue sample.

Screw cap tube containing approx. 1 cubic centimeter tissue, submerged in 70% ethanol.

EDTA blood samples in tubes; 5ml tube (top) and 2ml tube (bottom).

Dead animals

Samples should be taken in accordance with national legislation.

Tissue (max 1 cubic centimeter) from internal organ, skin or muscle. Placed in a plastic tube (2ml screw cap) containing 70% ethanol or frozen in a plastic bag.
Do not use formalin or methylated alcohol.

Serum (1-10 ml) in plastic tubes. Must be spun and seperated. Should only be provided if it is accompanied by a tissue sample.

CITES

Samples from some species will require CITES permits.

Within EU, according to CITES regulations there is no need for CITES export and import permits.*
Outside EU, then CITES export permits must be applied for at the national CITES office. Remember to apply for CITES export permits in due time before sending the samples.
CITES exemption is possible for scientific institutions (see article VII, §6 of the CITES convention). Your institution may apply. All four EAZA Biobank hubs have the CITES exemption.

When CITES export permits are obtained, please send a scanned copy to the contact person of the receiving Biobank hub, who will proceed to obtain CITES import permits.

* Exemptions may apply.

Shipping

Labeling

Label the sample with animal identifiers (transponder, ring, GAN or local ID), species name, tissue type and date when sample was taken.
Enclose the animal's ZIMS specimen report and contact details of sender, otherwise your sample will not be processed.
If you do not have a ZIMS report, then please include typed identification, species name, date of sampling and institution.

Storage

If you cannot ship the samples within 12 hours, then store them in the freezer until shipment is possible. Please ship the samples as soon as possible and please avoid arrival at the hub on a weekend.

Packaging

1 **Primary package**
The samples in tubes

2 **Secondary package**
Plastic container or bag for the samples with enough material within to absorb the total sample content.

3 **Tertiary package**
Reinforced envelope or cardboard box with ZIMS specimen report. Enclose an icepack in the envelope or box if the tissue samples are frozen.

Shipping

The package should be labeled on the outside with the diamond 'UN3373' logo and the text "Exempt animal specimen" and "refrigerate upon arrival".

The 'UN3373' label can be provided by one of the biobank hubs.

Biobank addresses

If you would like to send us your samples then please send them to the biobank hub relevant for your country.

Shipping country: **UK, Ireland, Qatar, UAE, Kuwait**

Edinburgh hub

ATT: **Dr. Helen Senn**
Address: Royal Zoological Society of Scotland
WildGenes Laboratory
134 Corstorphine Road
Edinburgh EH12 6TS, UK
E-mail: HSenn@rzss.org.uk

Shipping country: **Germany, Austria, Croatia, Czech Republic, Hungary, Poland, Russia, Slovakia, Slovenia, Switzerland, Ukraine**

Berlin hub

ATT: **Dr. Jörns Fickel**
Address: Department of Evolutionary Genetics
Leibniz Institute for Zoo and Wildlife Research (IZW)
Alfred-Kowalke Strasse 17
10315 Berlin, Germany
E-mail: fickel@izw-berlin.de

Shipping country: **Belgium, Luxembourg, The Netherlands, France, Greece, Israel, Italy, Turkey**

Antwerp hub

ATT: **Dr. Philippe Helsen**
Address: Centre for Research and Conservation
Royal Zoological Society of Antwerp
Koningin Astridplein 20-26
2018 Antwerp, Belgium
E-mail: Philippe.Helsen@kmda.org

Shipping country: **Denmark, Estonia, Finland, Latvia, Lithuania, Norway, Sweden, Portugal, Spain**

Copenhagen hub

ATT: **Dr. Christina Hvilsom**
Address: Copenhagen Zoo
Roskildevej 38
PO Box 7
2000 Frederiksberg, Denmark
E-mail: ch@zoo.dk

It is the responsibility of the lending institution to keep the biobank informed with their most up to date contact details.