EAZA Terrestrial Invertebrates Taxon Advisory Group

Best Practice Guideline for the noble chafer (*Gnorimus nobilis*)

By Signe Ellegaard & Eddie Bach. Copenhagen Zoo

EAZA Terrestrial Invertebrate TAG chair: Mark Bushell. Bristol Zoo Gardens

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TITAG Chair:

Mark Bushell, Bristol Zoo Gardens, Clifton, Bristol, BS8 3HA.

TITAG Vice-Chairs:

Tamás Papp, Chester Zoo, Moston Rd, Upton, Chester, CH2 1EU.

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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 "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. All forms/templates are available to download on the EAZA Member Area. 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.

Introduction

This Best Practice Guideline is based on information from the published literature on *Gnorimus nobilis* but primarily from our first-hand experiences working with the species both *in* and *ex situ.*

The noble chafer beetles are relatively short-lived and are therefore far from the typical zoo-animal. The adult beetles only live for a few weeks and *G. nobilis* is therefore not an obvious species to exhibit in a zoo. Nevertheless, it is a symbol of the strong insect decline we are facing, and it therefore plays the lead role in a conservation project of Copenhagen Zoo, aiming to secure the last few Danish populations and to restore their declining habitat. The larvae of this saproxylic species inhabit hollows created by wood-decaying fungi in dead or decaying deciduous trees such as *Quercus* sp. and *Fagus* sp. In contrast, they are primarily found in hollows in old traditional fruit orchards in England. Inside the hollows, the larvae forage on the wood mould and other accumulated organic matter, contributing to the nutrient recycling and wood decay. As a part of the conservation project, suitable trees are veteranized which speeds up the natural process of decay thereby creating new microhabitat for the species, but also for other species utilizing these hollows (such as woodpeckers, other saproxylic insects, bats etc.), is created. By that, *G. nobilis* functions as an umbrella species in the Noble Chafer Project of Copenhagen Zoo[1]. This Best Practice Guideline is therefore meant as a guide for keeping and breeding *G. nobilis* but may also serve as inspiration for breeding other saproxylic beetle species of conservation and/or education interest.

The current captive population in Copenhagen Zoo was founded by 1.2 individuals collected from the wild at one of the last two known localities in Denmark in 2016. By 2020, more than 3000 larvae have been bred over 3 generations and more than 2500 larvae have been reintroduce to the wild.

We would like to thank all our internal and external colleagues who have been and are still involved in our conservation project. A special thanks to Kristian Graubæk, Anton Asklund Johnsen, Marie Graversen, Matthew Smith, Deborah Harvey, Maria Fremlin, Yikai Zhang, and Rasmus Dylov. We want to thank the amazing photographers Henrik Egede-Lassen, Frank Rønsholt, and Steen Drodz Lund. Thank you to the staff of

[1] More information about the project can be found in Zooquaria #108.

Copenhagen Zoo, including Lene Vestergren Rasmussen, Maya Gaard Rasmussen, Jacob Riis, Martin Thy Jensen, Mads Frost Bertelsen, Mikkel Stelvig, and Bengt Holst. We would also like to thank our local landowners and sponsors.

Summary

This Best Practice Guideline covers a broad range of knowledge on the saproxylic beetle noble chafer *Gnorimus nobilis*. The beetle is relatively short-lived and is not a typical exhibition animal and the only captive population hold by EAZA institutions (at Copenhagen ZOO) is bred purely for *in situ* conservation purposes with related education and research activities. This document is meant as a general guideline for keeping and breeding *G. nobilis* specifically, but it may also be applicable for management of other similar saproxylic beetle species. The document is divided into three sections according to the EAZA template.

In Section 1, Biology and Field data, the current knowledge on the species' biology and field data is presented, including its conservation status and descriptions on its habitat, feeding behaviour and reproduction. The information presented in this section is based on the scarce published literature on the species as well as on our own field observations. For some of the subsections, information is based on observations from the captive population as field data in these areas is lacking.

Section 2, Management in Zoos and Aquariums, presents the best practice for managing the species which is relatively easy to keep and breed. The recommendations given are primarily based on our own experience as well as knowledge from breeders of a previous captive population in England. The most important aspect of keeping and breeding the species seems to be to use of the correct substrate, as this serves as the larval food source. The larvae do thus not need direct feeding and the animals can be left alone during most of the time. Furthermore, the environmental conditions such as temperature and substrate moisture seem to be of importance for successful development and reproduction. This section presents recommendations for enclosure design, feeding, breeding, and handling, among others. In the end, a list of suggested future research topics is given.

In the last section all references for the literature cited in this document are presented.

Contents

Section 1: Biology and field data

Biology

1.1 TAXOMONY

- Class: Insecta

- Order: Coleoptera
- Family: Scarabaeidae
- Sub-family: Cetoniinae (Flower chafers)
- Tribus: Trichiini
- Genus: *Gnorimus*
- Species: *G. nobilis*
- Sub-species: *Gnorimus nobilis macedonicus* (Baraud, 1992), *Gnorimus nobilis nobilis* (Linnaeus, 1758), *Gnorimus nobilis var. cuprifulgens* (Reitter, 1908)
- Common names: Noble Chafer (English), Grüner Edelscharrkäfer (German), Le Verdet (French), Grøn pragttorbist (Danish), Ädelguldbagge (Swedish), Zdobenec zelenavý (Czech), Зелёный пестряк (Russian), Zacnik (Polish)

The name *Gnorimus* comes from the ancient Greek where *gnõrimos* means 'famous'. *Nobilis* derives from Latin meaning 'noble' (Benisch, n.d.).

Earlier scientific names include *Scarabaeus auratus* (Shrank, 1781), *Cetonia cuspidata* (Fabricius, 1787), *Scarabaeus igneus* (Voet, 1769), and *Scarabaeus viridulus* (De Geer, 1774) (as refered to in Tauzin, 2000). In literature from 1994 to 2006 the genus *Gnorimus* (Le Peletier de Saint-Fargeau & Serville, 1828) might be referred to as *Aleurostictus* (Kirby, 1827). Krell, Smith, Ballerio, & Audisio (2006) argued to conserve the generic name *Gnorimus* and consequently the synonym *Aleurostictus* is not used anymore.

1.2 MORPHOLOGY

1.2.1 Weight & length / Measurements

No measurements for wild specimens are currently available. Therefore, all data given here are from captive individuals from the Copenhagen Zoo population. Due to lack of field data, size variations between geographic regions cannot be ruled out and the

measurements presented here might therefore represent animals of relatively small or large size compared to other populations. A digital scale with minimum 0.01 g precision is needed to obtain precise weights. Length of adults is most easily measured by photographing the individual on millimeter paper (preferable in a small see-though container with a lid and analyzing the measurements on the computer thereafter (e.g. using the software ImageJ). Length of larvae is measured directly using a ruler or similar by carefully stretching the larval body (which is usually curled in a c-form).

Eggs and larvae will increase their weight throughout development; however, larval weight is very variable during development with an increase from egg hatching until a peak before the weight decreases again when entering winter diapause. The eggs have 1-2.5 mm diameter with a weight below 0.02 g (fig. 1). Right after hatching, larval weight is less than 0.05 g and the maximum weight before entering winter diapause in the third and last larval stage is 1.3 g. Larvae reach up to 40 mm in length.

The larvae go through three larval stages before pupation (fig. 2; section 1.7) which can be determined from the width of the sclerotized head capsule (referred to as HCW) which stays constant throughout each of the larval stages (Daly, 1985). The larvae put on weight continuously through development, which is why larval weight cannot be used to determine which larval stage the specific larva is in (fig. 3). The most reliable method to determine the larval stage is to measure HCW directly (using a digital caliper (fig. 4) regularly to determine when the larval stage has change (see fig. 5) – or, for the trained eye, it can be determined visually by just looking at the size of the head capsule. If larvae are kept singly, the presence of a shed larval skin in the substrate can also be enough to determine that a change in the larval stage has taken place.

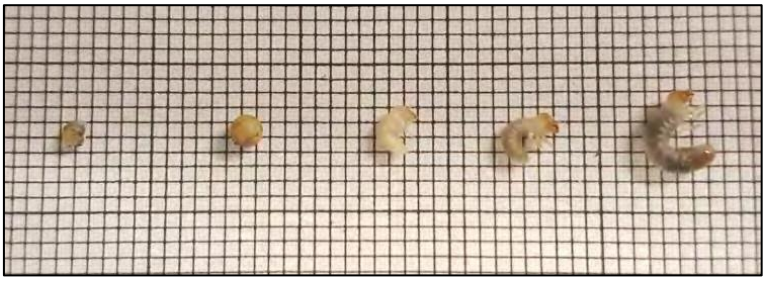

***Figure 1**. Eggs and newly hatched larvae in the first larval stage. The larvae in the middle has hatched within one hour, the next larva to the right has hatched within a few hours, and the last larva to the right has hatched within a few days.*

***Figure 2**. Larvae in the three larval stages. The larva in the bottom right is L1, the larva in the upper right is L2, and the larva to the left is L3.*

***Figure 3**. Two larvae both in the third and final larval stage. Notice the big size difference with a relatively large head compared to the body on the lower left larva that is newly molted. The upper right larva has been L3 for a longer time and has therefore put on more weight resulting in a relatively small head compared to the body.*

***Figure 4**. Measurement of HCW of a larva using a digital caliper.*

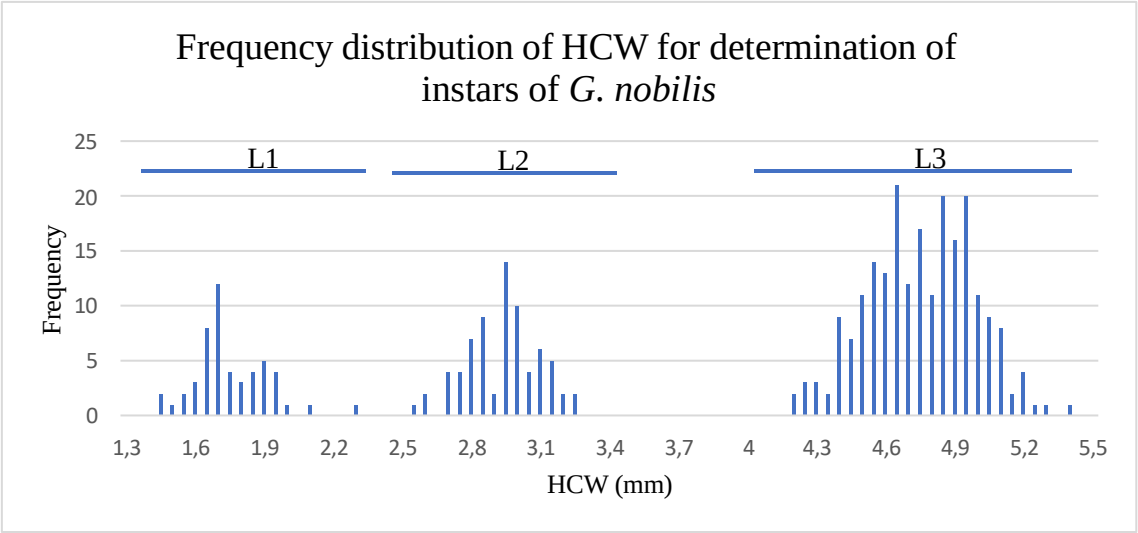

***Figure 5**. The frequency distribution of head capsule widths (HCW) measured on 341* G. nobilis *larvae using a digital caliper. Larvae with a HCW on 1.40-2.30 are assigned to L1; larvae with a HCW on 2.50-3.30 are assigned to L2; larvae with a HCW on 4.20-5.50 are assigned to L3.*

Adults are 1.5 to 2.0 cm long and weight between 0.3 to 0.6 g. No significant size or weight differences between sexes has been found for either larvae or adults.

1.2.2 Coloration

Eggs are pale white when laid and will gradually turn more yellow until hatching (fig. 1 and 8).

Larvae are pale white right after hatching but with the rear end of the body gradually turning more grey or brown when the larvae start eating (as the gut content is visible through the transparent skin). The head is pale white after hatching and after each molt but will start sclerotizing immediately and gradually get an orange to brown color (fig. 6). Through development, the larval body turns more yellow as fat is continuously build up until the onset of diapause. (fig. 7).

***Figure 6**. A larva in the process of ecdysis (see section 1.7.1). Notice the pale head and body.*

***Figure 7**. Two L3 larvae in different times of development. The larva to the right has built up more fat than the paler larva to the left.*

Pupae are pale white, gradually turning more yellow or orange with time (fig. 12 and 13). Right before eclosion a light green metallic sheen on the developing elytra can be seen.

The body color of adults is variable from primarily metallic green to brown, copper, or even gold, often showing iridescence (fig. 14). The dorsal side of the adult body may have small white spots on thorax, elytra, and abdomen. The ventral side of the body is darker than the dorsal side, usually black.

1.2.3 Description

Eggs are small and globe like (fig. 1 and 8).

Larvae (fig. 9) have a typical scarab larval c-form (often referred to as a “grub”) with visible spiracles along the body and well-developed mouthparts and thoracic legs. Prothoracic shield is visible just behind the head on each lateral side of the first segment. The body is firm to the touch and is covered with short very fine hair. The anal opening on the posterior end of the body is transverse. In the third larval stage, sexes can be

distinguished by the presence of Herold's organ on the ventral side of the males' body segment (above the anal opening (fig. 10 and 11)).

***Figure 8**. Eggs in the substrate.*

***Figure 9**. L3 larva.*

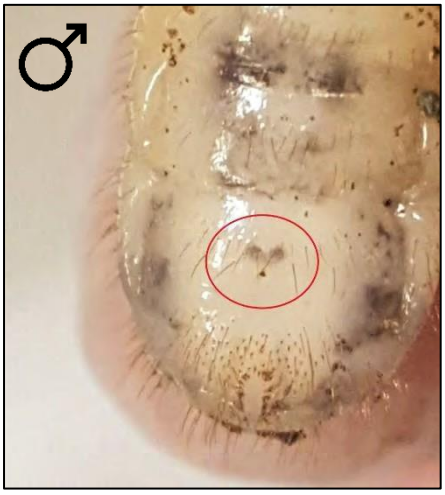

***Figure 10**. Male larva (L3). The Herold's organ is marked.*

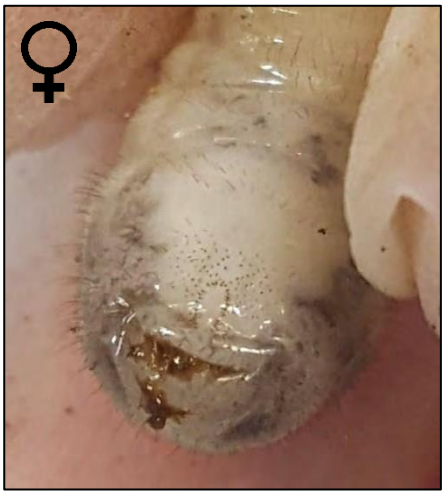

***Figure 11**. Female larva (L3). Notice the lacking Herold's organ.*

***Figure 12**. Pupa, earlier in development than in fig. 13.*

***Figure 13**. Pupa, later in development than in fig. 12.*

Imagoes have long legs and a more or less typical scarab form and size. The elytra (hardened forewings) may be covered with small wrinkles. The protonum (the hard plate covering the front part of the thorax dorsally) is narrower than the elytra which do not reach the posterior end of the abdomen. Especially the "waist" between protonum and elytra differentiates it from the rose chafer *Cetonia aurata* which it is often confused with. Scutellum (the small triangular plate between the forewing bases) forms a small equilateral triangle.

Sexes can be distinguished by the look of the pygidium (the posterior tip of the abdomen) which has a notch in females but not in males. Furthermore, in Western Europe, the middle segment of the mesotibia (the middle leg) is thicker in males than in females (fig. 15).

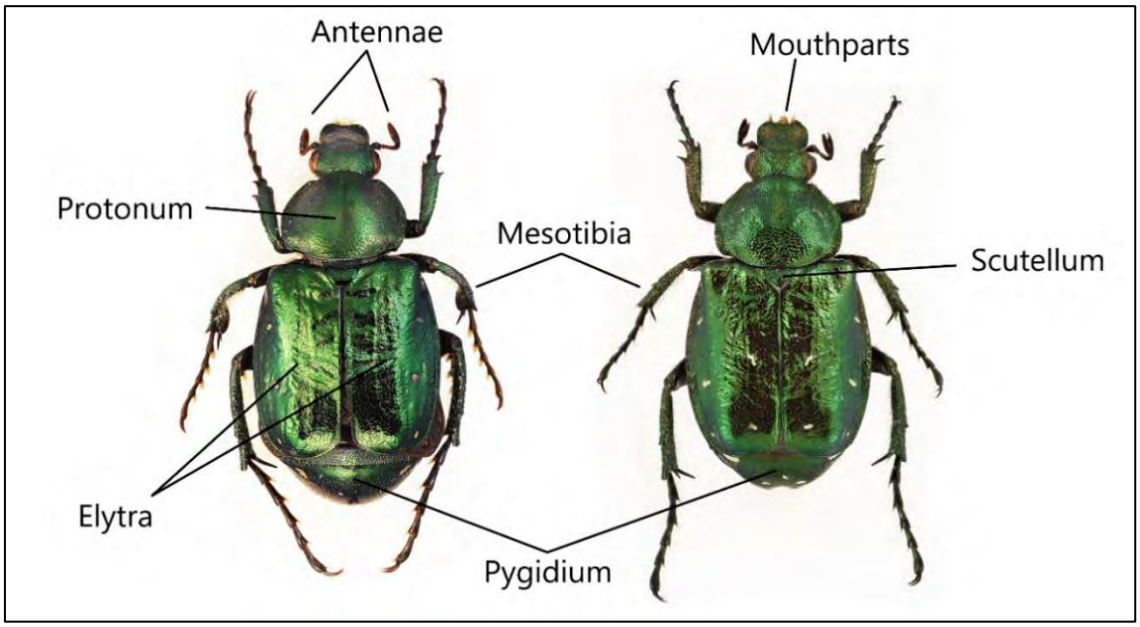

***Figure 14**. Adult beetles with relevant morphological terms as referred to in the text. Male is shown on the left, female on the right. Modified from photos by Siga (www.creativecommons.org/licenses/by-sa/3.0/).*

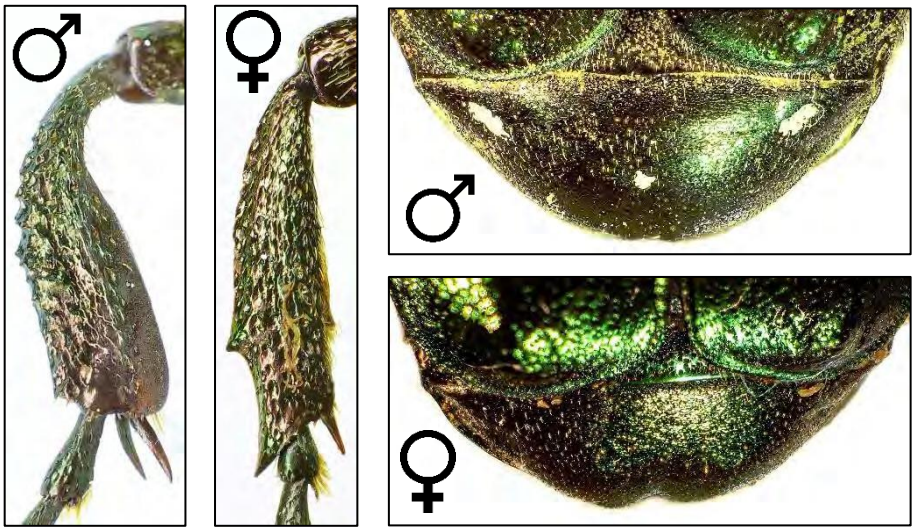

***Figure 15**. Sexual differences on the mesotibia and pygidium. Photos by Siga (www.creativecommons.org/licenses/by-sa/3.0/).*

1.3 PHYSIOLOGY

As in many other insect species, the larvae enter diapause in the cold winter months to enhance survival in this unfavorable time (Tauber & Tauber, 1976). The winter diapause, a state of dormancy which is presumably initiated mainly by the seasonal changes in temperature and photoperiod, is expressed by several features, including physiological adaptations such as lowered metabolism and developmental arrest (Koštál, 2006). Leading up to the onset of the winter diapause, the larva will stop eating and empty its gut (fig. 16) (resulting in a loss of weight), leaving the previously brownish/blackish hind end of the body pale and "shrunken". To survive the low winter temperature, the larval survival strategy is freezing-avoidance rather than freezing-tolerance (Vernon & Vannier, 2001; Vernon, Vannier, & Luce, 1996). By clearing the gut, the larva removes the nucleation source making it possible that the body fluids can be cool below its freezing point without changing into a solid phase (Bale, 1993). This ability is known as supercooling and helps the larva to avoid freezing of the body fluids in winter.

***Figure 16**. A dissected* G. nobilis *L3 larva during winter diapause. Notice the empty gut.*

Except for the two studies on the freezing-susceptibility of the larvae and one on body water loss in larvae (Renault, Vernon, & Vannier, 2005), no information on species-specific physiology is currently available. For more information on general internal morphology of Scarabaeidae larvae see Rapp (1947) and Tashiro (1990).

1.4 LONGEVITY

The lifespan of adults is relatively short, varying in both captivity and in the wild from 4-6 weeks.

The total lifecycle varies from one to three years depending on environmental conditions, however two years seem to be most common in the published literature (see for example Alexander & Bower, 2011; Owen, 1989; Pawlowski, 1961). At Copenhagen Zoo (in the following referred to as CPH ZOO), the first generation took two years to develop whereas the following generations only used one year.

Field data

1.5 CONSERVATION STATUS/ZOOGEOGRAPHY/ECOLOGY

1.5.1 Distribution

G. nobilis is endemic to Europa and is found in most regions except of the far north. Its distribution is patchy and very localized in several countries e.g. in the UK, Sweden, and Denmark. The countries in which the species can be found include: Albania, Andorra, Austria, Belarus, Belgium, Bosnia and Herzegovina, Bulgaria, Croatia, Czechia, Denmark, Estonia, France (mainland and Corsica), Germany, Greece (mainland), Hungary, Italy (mainland), Latvia, Liechtenstein, Luxembourg, Moldova, Montenegro, Netherlands, North Macedonia, Norway, Poland, Portugal (mainland), Romania, Russian Federation (Kaliningrad, South European Russia, Central European Russia), Serbia, Slovakia, Slovenia, Spain (mainland), Sweden, Switzerland, Ukraine, United Kingdom (Great Britain) (Mannerkoski et al., 2010).

1.5.2 Habitat

G. nobilis is an obligate saproxylic species being depended on dead or decaying wood for development. It is found in standing trees that are still alive but have cavities where the larvae can develop in the wood mould (i.e. wood that has been attacked/infected by wood-decaying fungi such as tinder fungus *Fomes fomentarius*, sulphur polypore *Laetiporus sulphureus*, *Tramestes* sp., and in the UK also *Phellinus pomaceus*) (fig. 17). In Hungary, larvae have also been found in dead lying trunks (Mannerkoski et al., 2010).

***Figure 17**. An old decaying tree with cavities for* G. nobilis

***Figure 18**. Open grown forest in Denmark, a typical habitat for* G. nobilis *in large parts of its distribution range.*

***Figure 19**. Old apple orchard, the typical habitat for* G. nobilis *in England. Photo by Sarah Smith (creativecommons.org/licenses/by-sa/2.0/deed.en)*

The species is dependent on old open-grown forests in the western part of the species distribution range (fig. 18) but requires more shade under more continental conditions in its eastern distribution. The tree species seems not to be of great importance, the determining factor making the trees suitable for the species may be the level/stage of wood decay. In continental Europe, the larvae have been found in *Salix* spp., *Fagus sylvatica*, and *Quercus* spp. among others. In England, the species seems more restricted to species of orchard fruit trees (such as plum *Prunus domesticus*, apple *Malus*, pear *Pyrus*, and cherry *Prunus* spp.) (fig. 19) as well as *Quercus* spp., *Betula* spp., and *Crataegus monogyna* (Mannerkoski et al., 2010; Uff, 2019; Whitehead, 2002).

As a food source for the adult beetles, blooming flower heads of e.g. umbellifers no more than 700 m from the breeding trees are required (Whitehead, 2002).

Weather conditions in the species' distribution area are characterized by a general temperate climate. Summers are cool to hot and dry (with average temperatures ranging from just below 20°C in the northern areas and 25°C in the south) and cold to mild winters (with average temperatures ranging from the freezing point to up to 12°C). The average number of days with precipitation per year ranges from approx. 50 days in the south to 100-150 in the north.

1.5.3 Population

There is currently no data available on either the total population or national/regional population sizes.

1.5.4 Conservation status

G. nobilis is classified as Least Concern with stable populations trends on the IUCN Red List (latest assessment in 2009, but with an annotation stating that the assessment "needs updating") (Mannerkoski et al., 2010). However, there have been declines reported from several countries, especially in the north-western parts of the species' distribution range. National and regional classifications are provided in Table 1 for areas where these are available.

Table 1 – National (or regional) conservation status for *G. nobilis*

Country (- region)	Category	Year of assessment	Reference
Albania	CR	2013	(Republic of Albania Ministry Environment Forests and Water Administration, 2013)
Belgium - Flanders	CR	2015	(Thomaes, Drumont, Crevecoeur, & Maes, 2015)
Bosnia and Herzegovina	LC	2013	(Đug, 2013)
Czech Republic	VU	2017	(Nature Conservation Agency of the Czech Republic, 2017)
Denmark	EN	2019	(Aarhus Universitet DCE - Nationalt Center for Miljø og Energi, 2019)
Estonia	DD	2008	(Commission for Nature Conservation of the Estonian Academy of Sciences, 2008)
France	LC	2013	(Inventaire National du Patrimoine Natural, 2013)
Germany	3 (VU)	1998	(Binot, Bless, Boye, Gruttke, & Pretscher, 1998)
Italy	NT	2015	(Carpaneto et al., 2015)
Norway	NT	2015	(Artsdatabanken, 2015)
Russia - Kaliningrad	VU	2018	(Alekseev, 2018)
Sweden	NT	2020	(SLU ArtDatabanken, 2020)
Switzerland	NT	2016	(Monnerat, Barbalat, Lachat, & Gonseth, 2016)
United Kingdom	VU	2016	(Lane & Mann, 2016)

The general status of saproxylic beetles in Europe is evaluated in the European Red List of Saproxylic Beetles (Cálix et al., 2018). Out of the 688 assessed species, 17.9% have

been classified as threatened (CR, EN, or VU), but for close to one quarter of the assessed species, their risk of extinction could not be evaluated due to lack of data (DD).

Quantity and quality of dead and decaying wood as well as tree density, and habitat continuity are some of the major factors determining saproxylic beetles' richness and diversity. The major threats to the wild populations of saproxylic beetles (including *G. nobilis*) therefore include logging, tree loss (i.e. threats of tree age structure gaps, loss of ancient and veteran tress), wood harvesting and agricultural intensification and development (Cálix et al., 2018). The lack of old and hollow trees in forests in Europe is increasing, mostly due to extensive commercial forest management, where old and non-valuable (economically) wood, including deadwood, is removed from the forests leaving very little suitable habitat for the saproxylic forest species (Lindenmayer et al., 2014). Likewise, non-, or minimum-intervention management systems where grazing by large herbivores is excluded can lead to canopy closure and loss of ancient trees which has negative impacts for the species. For example, canopy overgrowth will cool down the wood mould thereby slowing larval development (Mannerkoski et al., 2010).

1.6 DIET AND FEEDING BEHAVIOUR

1.6.1 Food preference

Larvae are saproxylophagous eating heartwood rot and the accumulating debris in the tree cavity created by heartwood-decay fungi. The tree species does not seem to be of importance, and in England, the larvae are also found in old fruit orchards as mentioned in section 1.5.2 (Alexander & Bower, 2011). Exactly what the larvae extract from the wood mould is not known but it has been proposed that cellulose-digesting bacteria are present in their gut. It could also be that they digest micro-organisms from the wood mould (Alexander & Bower, 2011). Larvae of *Cetonia aurataeformis* are known to be able to decompose lignin (Micó, Juárez, Sánchez, & Galante, 2011) but this does not seem to be the case for *G. nobilis* as their fecal pellets are composed mainly of lignin (Whitehead, 2002) (indicating that they are not able to digest this).

Like *Osmoderma* larvae, *G. nobilis* larvae may be dependent on a diet containing not only the wood itself but also on other organic matter accumulating inside the tree hollows (for example leaf litter and fecal pellets) (Landvik, Niemelä, & Roslin, 2016). A feeding

experiment has led to the conclusion that they can live of pure frass of their own as well as of different fungal substrate such as oak sawdust infested by *Laetiporus sulphureus* or *Trametes versicolor*, at least for a few weeks. However, whether these are preferred food sources in the field is not known. Larvae have also shown to be able to consume food items of animal origin; skeletal fragments of beetles have been found in their feces (Whitehead, 2002).

Adults are nectari- and palynivorous feeding on pollen (protein source) and nectar (carbohydrate source) from blooming flowerheads of various plants. In Denmark, adults have been observed feeding on elder (*Sambucus nigra*), ground elder (*Aegopodium podagraria*)), dog rose (*Rosa canina*), hemp-agrimony (*Eupatorium cannabinum*), guelder-rose (*Viburnum opulus*) with elderflower being the most common (pers. obs.). In England, species such as meadowsweet (*Filipendula ulmaria*) and hogweed (*Heracleum* sp.) are mentioned as preferred food items.

1.6.2 Feeding

Larvae feed continuously throughout the day on the substrate in which the live, continuously filling the cavity with small fecal pellets (frass). The frass is relatively soft when laid but quickly turns into small hard pellets (fig. 27). The larvae do not feed when in winter diapause. According to Pawlowski (1961), the larvae feed for 40-44 weeks (depending on environmental conditions) during a calendar year.

Adults are observed feeding in the daylight hours on warm, sunny days. Some might not feed at all but stay inside the tree cavity in which they eclosed if they have already mated and the substrate quality is good enough to lay eggs in.

1.7 REPRODUCTION

Information given here is from observations on the captive population in CPH ZOO. However, it is expected that similar patterns are seen in wild populations as well.

1.7.1 Developmental stages to sexual maturity

G. nobilis eggs hatch within one-two weeks. The species undergoes three larval stages (fig. 2 and fig. 20) (average developmental time for L1 is 9-15 days; for L2 21-28 days; and for L3 approx. 9 months) before pupation. The molting from one stage to the next (ecdysis) is characterized by the shedding of the larval skin (the cuticle), including the highly sclerotized cuticle on the head (fig. 6). Immediately after shedding of the cuticle, the larval body (which has now increased in size) is soft and pale but quickly the cuticle hardens, and the head is once again sclerotized. After the third and last larval stage, the larva molts into the pupa and after approx. one month, the imago ecloses. The sexually mature imago will eclose from the pupa approximately 10-11 months after the egg was laid. The duration of the developmental stages seems to be temperature dependent (with faster development at increasing temperature) and most likely also dependent on humidity and food quantity and quality.

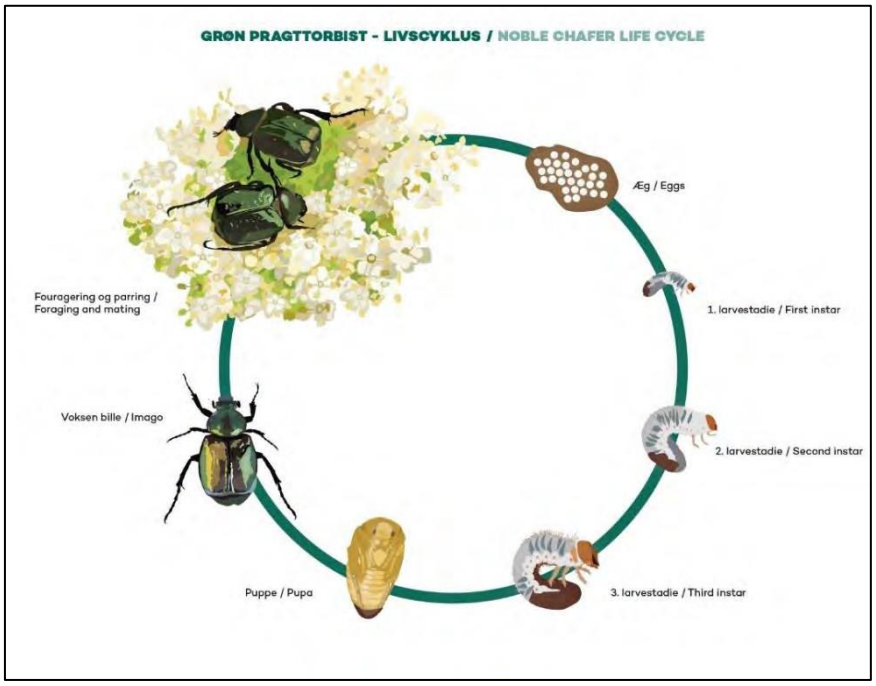

***Figure 20**. Graphical representation of the life cycle.*

1.7.2 Age of sexual maturity

Imagoes will eclose after 1-3 years as larvae, depending on how favorable the conditions are. Presumable, imagoes are sexually mature instantly after emergence (or they might need to feed before reaching sexual maturity as with *C. aurata* (Maria Fremlin, pers. comm.)).

1.7.3 Seasonality of cycling

Mating and egg laying happen in spring/summer right after adult emergence (a few days after eclosion) (in Denmark around May-June, in England around June-July). Larvae develop during summer and early fall before entering the winter diapause in October-November and will either pupate the following spring (April-May in Denmark) or continue larval development through the following year before pupating next spring. Whether the larvae overwinter once or twice (i.e. as the second or third instar) must depend on the local microclimatic conditions as well as the date of oviposition. The life cycle including seasonality for both the one- and two-year cycling is depicted in Table 2 and 3 (pers. obs.; Matthew Smith, pers. comm.).

***Table 2** – The two-year life cycle of* G. nobilis*. The larvae will overwinter as L2, continue feeding and develop into L3 the following spring, and thereafter overwinter one last time as L3 before pupating the following spring.*

	Month											
	J	F	M	A	M	J	J	A	S	O	N	D
Year 0						Egg						
							Larva L1, L2			Winter diapause		
Year 1			Larva L3							Winter diapause		
Year 2												
				Pupa								
					Imago							

***Table 3** – The one-year life cycle of* G. nobilis*. The larvae will develop into L3 during its first autumn, enter the winter diapause and pupate the following spring.*

	Month											
	J	F	M	A	M	J	J	A	S	O	N	D
Year 0						Egg						
							Larva L1, L2, L3			Winter diapause		
Year 1												
				Pupa								
					Imago							

1.7.4 Clutch size

The female lays 25-50 eggs over her lifetime.

1.8 BEHAVIOUR

1.8.1 Activity

Larvae are presumably active throughout the day (as they are not exposed to sunlight and thus not affected by the photoperiod). The larvae enter winter diapause in around October to March where they are inactive. According to Pawlowski (1961), larvae are active 40-44 weeks a year, leaving 8-12 weeks for the diapause. Our experience reveals a longer diapause period on approximately 16 weeks.

Adult beetles are diurnal, and activity (foraging and mating) have mostly been observed on warm, sunny days. Where the adults are at night is not known and further fields observations would be required to answer this.

1.8.2 Locomotion

Larvae crawl with the dorsal side up (unlike *C. aurata* larvae which move with the ventral side upwards (Fremlin, pers. comm.). Locomotion seems to happen with both the legs and movement of the whole body.

Adult beetles move from breeding site to/from feeding site by flying. The flight is almost soundless (as opposed to the flight of *C. aurata* adults which are quite noisy especially in take-of (pers. obs.)). Adults may also move around by crawling for short distance movement (e.g. on a flower head while foraging and when mating).

1.8.3 Predation

No reports in the literature have been made of eggs, larvae, pupae, or adults being predated on by other species. However, it is highly likely that they are eaten (in all developmental stages) by other animals known to predate on similar beetle species inhabiting the same habitats. This could include predation by other invertebrates (such as larger beetles and ants), reptiles (such as lizards), birds (such as woodpeckers and

corvids), and mammals (such as wild boars, bears, foxes, shrews, and badgers) (Chiari et al., 2014; Ulyshen, 2018; Whitehead, 2002).

Larvae have been observed killed in captivity by a lesser stag beetle *Dorcus parallelipipedus* hiding in the substrate. Furthermore, cannibalism has been observed in larvae in captivity, especially after they reach the second larval stage (presumably due to overcrowding, see section 2.3.1 for more information). In larval feces, skeletal fragments of beetles have been found, indicating that they themselves may consume food items of animal origin other than conspecific larvae (Whitehead, 2002).

1.8.4 Social behaviour

The species is not known to exhibit any special social behaviour. Larvae might be able to stridulate, as many larvae of Cetoniinae possess a stridulatory area on the mandibles and maxilla with stridulatory teeth which can be rubbed together to create a sound (Görres & Chesmore, 2019; Maurizi et al., 2017). This potential stridulation could be used for communication between conspecific larvae; however, this has not been studied in detail, especially not for *G. nobilis*.

1.8.5 Sexual behavior

As many species of Scarabaeidae, *G. nobilis* uses pheromones for sexual communication. A recent study by Harvey et al. (2018) revealed that the species shows an unusual pheromone-mediated behaviour where both sexes produce the pheromone, but only the males are attracted to it. The females in the study were repelled by the pheromone. This could be because the pheromone aids the dispersal of mated females by signaling where a high number of individuals are present thus helping the females avoid high-density breeding sites (and thereby increasing larval survival). The pheromone may furthermore aid the males to distinguish conspecifics from other similar Cetoniinae species.
The same study has found that the species-specific sex pheromone can be synthesized in the laboratory and this has shown to be an effective tool for monitoring the beetles in the field using pheromone traps.

For information on mating behaviour, see section 2.4.1 for observations from captivity.

Section 2: Management in Zoos and Aquariums

To our knowledge, captive breeding of noble chafers has been limited. A captive population was bred by Owen (1989; 1993) who published his observations on this. Matthew Smith took over that same colony from Owen and reared the species from 1999 to 2012, and in CPH ZOO a colony has been reared since 2016. Furthermore, Pawlowski (1961) and Vernon & Vannier (2001) have collected data on captive groups of *G. nobilis* but as parts of comparative studies on multiple species. The information given in this section is therefore primarily based on our own experiences with rearing the species at CPH ZOO. The breeding setup was initially based on Matthew Smith's experiences and has later been updated and adapted.

It is important to note that this information given here might not be the only strategy but has led to successful breeding over four generations so far.

2.1 ENCLOSURE

2.1.1 Boundary

Larvae are reared in plastic boxes with lids (fig. 21). A few small holes are drilled in both sides of the box just below the lid for ventilation (not in the lid, as heavy rain may flood the boxes if kept outdoors without further cover and because it makes it possible to stack the boxes on top of each other without blocking the air holes). Boxes and lids should preferably be black to minimize the light penetration as larvae naturally inhabit microhabitats where little or no light is present.

Adult beetles can be kept in similar plastic boxes but these boxes (or at least the lids) should be transparent to allow light inside the boxes. The lid is necessary to prevent beetles from flying away/escaping – however the beetles kept in captivity (especially indoors) do not seem to attempt to fly even when the lid is removed from boxes. Recently wild-caught beetles seem more agile and caution should be taken when removing the lid and when handling the beetles to avoid escape.

Larvae could also be reared in wooden nest boxes specially made for saproxylic beetles (see for example Hilszczański, Jaworski, Plewa, & Jansson (2014) and Jansson, Ranius, Larsson, & Milberg (2009)). The nest box should be kept in a cage with solid or fine

meshed boundaries to prevent escape of beetles when they emerge from the nest boxes after eclosion (fig. 22).

***Figure 21**. Black box with black lid used for rearing of larvae.*

***Figure 22**. Nest box used for rearing larvae.*

Because the adult beetles are relatively short-lived (4-6 weeks), the species have not been exhibited. A temporary terrarium design for exhibition could constitute of a glass terrarium (50 x 50 x 70 cm) with furnishing and maintenance as described in 2.1.3.

2.1.2 Substrate

The larval substrate contains a mix of large lumps of decaying wood (of e.g. *Quercus* or *Fagus* sp.), more finely crumbled rotted wood (like sawdust), and preferably some leaf litter or similar. It seems to be more the decomposition stage of the wood that is important for larval development (as food source) rather than the tree species. Larvae will either stay in the fine substrate or chew their way into the larger wood lumps making hollow galleries (fig. 23 and 24). The wood lumps should be decayed to a stage where the wood feels quite soft and can relatively easy be broken up by hand (or at least with a tool). If the wood lumps are too hard and "fresh", the larvae will not (probably because they cannot) chew in it.

***Figure 23**. Wood galleries made by the chewing action of the larvae.*

***Figure 24**. Wood galleries made by the chewing action of the larvae. Notice the larvae inside the galleries.*

With time, the larval excrements (in the form of fecal pellets measuring 1 x 2 mm, often referred to as frass) will build up in the substrate which therefore might need to be changed or topped up with fresh substrate, especially if many larvae are kept together in relatively small containers (fig. 25 and 26).

The substrate for adult beetles can be similar. Adults will either stay on top of the substrate when foraging or mating or they will dig their way into the fine substrate e.g. when searching for shade or when the females lay their eggs. Bigger wood lumps are not necessary for the adults, but if the offspring of the adults are not intended to be moved

***Figure 25**. Larger amount of larval frass in the substrate.*

***Figure 26**. Close up photo of larval fecal pellets.*

after egg hatching, the substrate should of course be prepared for the larvae from the beginning.

The fine substrate needs to be moist but with no water dripping from it when squeezed. It should not be as moist as substrate used for breeding of more exotic beetle species (Matthew Smith, pers. comm.) If the substrate is too dry and dusty, pupae might die and adult females will not lay eggs in it (Fombong, Haas, Ndegwa, & Irungu, 2012, pers. obs.). Similarly, if the substrate is too moist, the larvae might drown.

Make sure that there is no unwanted species present in the substrate before offering it to especially larvae and pupae by freezing it for 24 hours before use (se section 1.8.3 for potential predators).

2.1.3 Furnishings and Maintenance

For larvae, nothing but the substrate as described in 2.1.2 is needed. Adult beetles will crawl around on sticks, wood lumps, flowerheads etc. if offered (fig. 27).

***Figure 27**. Furnishing of box for keeping adult beetles.*

For both larvae and adults, the substrate is kept moist by spraying water (with a portable pressure sprayer) when the substrate seems dry. On summer day this may be once a day, during winter this may be only once a month. See section 2.1.2 for appropriate substate moisture.

2.1.4 Environment

Larvae should be kept at temperatures which correspond to the natural outdoor conditions. The temperature at CPH ZOO indoors is set to a summer temperature at approx. 24°C degrees (mid-April to end-October). To initiate the onset of winter diapause for the larvae, the temperature is lowered during the colder months to approx. 12°C degrees (from end-October to mid-April). Similarly, to stimulate termination of the diapause and to stimulate pupation (or resumption of feeding), the temperature is increased again in April. The temperature could also gradually be increased before April (and similarly be decreased gradually before October) to resemble the seasonal temperature fluctuations more naturally. A small waterproof temperature data logger is very useful for monitoring the temperature in the substrate (e.g., HOBO® Pendant® MX2201 logger). Data loggers with humidity recording would also be highly useful for monitoring the environment.

Larvae can be kept in boxes outdoors if the climate is similar to that of the species' distribution area. However, make sure that plastic boxes used for the larvae are protected during winter, e.g. with an insultation blanket wrapped around the boxes, to avoid fatal freezing (although the overwintering third-instar larvae might survive temperatures down to minus 19.8°C degrees (P. Vernon & Vannier, 2001)).

High temperatures often lead to faster developmental rates (until reaching a certain maximum thermal threshold) but sometimes also to lower survival and lower breeding success (Danks, 2000). This is also what we at CPH ZOO have found for *G. nobilis:* they seem to thrive best when kept outdoors, although growth and development seem faster indoors. It is therefore desirable to keep the larvae at a natural temperature range to ensure the greatest survival probability and reproductive success.

For larvae, the photoperiod does not seem to make a big difference as the larvae naturally stay inside the tree hollows where there is little to no light penetration. However, it may

be important to ensure that the larvae are not exposed to too much light for longer periods at a time as this may disturb them. The photoperiod indoors at CPH ZOO has been kept at 12h:12h (L:D) when the temperature is approx. 24°C and 5h:19h when the temperature is approx. 12°C.

2.1.5 Dimensions

Larvae are reared in lidded plastic boxes measuring 35 x 35 x 45 cm ≈ 55L (or larger) filled 3/4 to the top with substrate. It is our impression that the maximum number of larvae per space unit depends on which developmental stage the larvae are in; however, this exact number of larvae is not known. At CPH ZOO, a density of 50 larvae in a 55L box works well. It is important to note, that the more larvae are kept together (independent of the developmental stages), the more often substrate needs to be topped up or changed, as frass will continuously accumulate and replace the substrate and thereby the larval food source.

In plastic containers measuring 13 x 14 x 15.5 cm (fig. 28), up to 10 larvae have been reared together, but the mortality of newly eclosed adults seems relatively high in these setups (up to 50%). Successful rearing of single larvae has been done in small round containers with a diameter on 10 cm and 4 cm height as well as in petri dishes with 9 cm diameter and 1.5 cm height (for the purpose of systematic data collection and easy observations on changes in developmental stages; see section 2.4.4 for more information regarding this) (fig. 29).

***Figure 28**. Small container for rearing of up to 10 larvae.*

***Figure 291**. Petri dishes made ready for single larvae.*

Adult beetles are kept in similar plastic boxes of 55 liters, but only filled halfway to the top with substrate. Successful breeding has been observed with up to 30 individuals in one box but would probably take place with even more beetles kept together.

2.2 FEEDING

2.2.1 Basic Diet

Larvae feed on most types of soft, decaying wood of deciduous species such as *Quercus* spp., *Fagus sylvatica*., *Betula* spp., *Malus* spp., and *Prunus* spp. which constitute their substrate. Both white rot and red rot is accepted by the larvae. They will also feed on larval frass as well as other compostable material. They may furthermore feed on items of animal origin, such as dead adult beetles. See section 1.6.1 and 2.1.2 for more information on larval diet and feeding behaviour.

Adults will feed on pollen (protein) and nectar (carbohydrates) from mostly white flowerheads (see section 1.6.1 for a range of species). At CPH ZOO, they are given blooming elderflower or guelder rose (depending on what is in bloom at the given time) and "beetle jelly". At CPH ZOO, beetle jelly with banana (which seems to be most attractive to the beetles), apple, melon, or strawberry flavor is used.

2.2.2 Method of Feeding

Food items are available at all times. The larvae will feed from their substrate which is to be topped up/replaced when necessary.

Adults are offered a few branches with blooming flowerheads as nectar and pollen source. The stem is cut and placed in water in a small plastic cup with a lid where a small hole has been drilled (for the stem to be placed through) (see fig. 27). It is important to ensure that the hole in the lid is not bigger than the stem; the stem should fit just through the lid to ensure that the beetles cannot enter the water and drown. The cup with the flowerhead is placed directly on top of the substrate. The flowerhead is replaced with a fresh one when it starts to wither. The beetle jelly is placed on top of the substrate as well and is replaced when empty or when it begins to rot. A few fresh flowerheads supplemented with a small container of beetle jelly is enough for 30 adults for a few days.

2.2.3 Water

It is not necessary (nor recommended due to the risk of drowning) to provide an open water source. Adult beetles get water from nectar and from the small water droplets that are formed on branches and wood lumps after spraying water.

2.3 SOCIAL STRUCTURE

2.3.1 Basic Social Structure

Successful breeding has been achieved by keeping up to 15 female imagoes together with 15 males. However, breeding has also taken place when just one of each sex has been kept together. Other constellations might work just as well and no constellations have been observed to have a negative effect on breeding and the welfare of the animals, Therefore, breeding will probably also take place if more individuals are kept together.

Be aware of potential overcrowding of the larvae once the eggs hatch if these are not separated in smaller groups (as each female will lay up to 50 eggs). Overcrowding can have inhibitory effects on development (Danks, 2000) and can furthermore lead to cannibalism (especially when larvae have reached the second instar). Therefore, larvae should not be kept together in too large amounts. We recommended keeping no more than 50-100 larvae per 55L box.

2.3.2 Changing Group Structure

The group structure can be mixed and changed without any problems if overcrowding is avoided. If needed, newly emerged beetles can be transferred to other boxes. There has not been observed any aggression when beetles are moved around.

2.3.3 Sharing Enclosure with Other Species

There have been no trials of keeping *G. nobilis* together with other species. It is however recommended that they (in any developmental stage) are not held together with other burrowing and/or carnivorous species to prevent disturbance and potential predation. We have seen predation on *G. nobilis* by a lesser stag beetle imago (*D. parallelipipedus*) which was accidentally kept together with 30 L3 larvae and assumingly caused the death

of more than half of them. No specific advantages are known to keeping *G. nobilis* together with other species in captivity.

2.4 BREEDING

In CPH ZOO, one group of *G. nobilis* is kept indoors (under the conditions as described in section 2.1.4) and one group is kept outdoors under natural conditions. Both groups follow the life cycle seasonality which is reported for wild populations in the literature.

2.4.1 Mating

Adult beetles eclose from the pupae in May-June and seem to be sexually mature right after emergence. The beetles will therefore pair and mate immediately after being mixed. Similarly, if a male and a female emerge in the same box at the same time, they will have mated within the first 24 hours (and probably long before that). If mating needs to be controlled, the specific larvae should be kept singly before pupation, so that beetles do not unintentionally mate after emerging from the same substrate before they are discovered and moved as required.

Copulation can be observed taking place on top of the substrate, on wood lumps, or on flowerheads. The male mounts the female from behind using his legs (including his broad mesotibia) to hold on to her. Fertilization is internal, with the male extending and inserting the aedeagus (fig. 30) into the female (fig. 31). Males will often be observed to grasp and hold on to the females even though direct copulation does not place (fig. 32).

***Figure 30**. The male aedeagus extended from the pygidium (on a dead male).*

***Figure 31**. Copulation with the female at the bottom and the male on top with the aedeagus inserted in the female's genital opening.*

***Figure 32**. Male grasping on to a female but without direct copulation taking place.*

Successful breeding has been observed with both a 1:1 male:female ratio, but also with lower and higher ratios. In theory, there should be more females than males in a breeding box to obtain the highest number of offspring, as it seems like females only mate once, whereas males will mate several virgin females if available (indicating a polygamous mating system).

2.4.2 Egg Laying and Incubation

The females will lay the first eggs shortly after mating (within the first 24 hours). The eggs are laid under the surface of the substrate, and the female will happily lay them in the same substrate as the beetles are eclosed in, if suitable (see section 2.1.2). One female will typically lay between 20-50 eggs over the course of 3-4 weeks and will die thereafter (fig. 8). The eggs are small and pale when laid and it is therefore advised to leave them *in situ* until they have hatched 1-2 weeks later. Be very careful with handling (see section 2.6.3 for advice). The eggs should be kept under the surface of the substrate which should not be too dry and dusty nor too moist.

2.4.3 Birth/Hatching

1-2 weeks after the eggs are laid, they will hatch in the substrate. However, please note that not all eggs might hatch, maybe because they have not been fertilized or due to an unsuitable substrate. The hatching process has been observed to last no longer than a few

hours. The first instar larvae are very small and pale right after hatching (fig. 4). Caution must be taken if handling or movement of the newly hatched larvae is necessary (see section 2.6.3 for advices on handling).

2.4.4 Development and Care of Young

No parental care takes place in the species. Once the eggs are laid, the female will not interact with or care for them.

The larvae will go through three instars from egg hatching in May-June to pupation in March-April. The larvae will stay under the substrate surface or inside the wood lumps all the time. As long as temperature and substrate quality and quantity are maintained the larvae can be left alone in the substrate. Only if you wish to control and/or monitor the population the animals need to be handled (see section 2.6.2 for more on this).

As described in section 2.1.4, the larvae will feed until the weather begins to cool (or the temperature is decreased if reared indoors) in September-October. They will empty their gut at this point, indicated by a weight loss and a change of body coloration, and enter winter diapause. It is recommended that the larvae are not disturbed too much at this point. They will then start feeding again in spring and if they entered the diapause as L2 they will overwinter once more. Or, if they entered the diapause as L3, they will pupate in spring (after gaining some weight again (P. Vernon & Vannier, 2001)).

Pupation will either take place under the substrate surface (fig. 33) or inside the wood lumps (fig. 34). No strategy seems to be preferred over the other and what determines where the larvae will pupate needs further investigation. If staying in the substrate, the larvae will make a pupal cell in which it will pupate. This cell is very fragile and cannot be moved in the same way a cell of a *Pachnoda* species or a cocoon of a rose chafer can be moved. If this chamber collapses (due to disturbance from e.g. handling or overcrowding of larvae), development is unlikely to continue successfully (Matthew

***Figure 33**. Pupae in the fragile cells in the substrate "attached" to the side (here plexiglass) of the box. Notice the heads directed upwards in the substrate and the shed larval skin in the bottom of the cells.*

***Figure 34**. A chamber inside a wood lump in which a larva pupated. Here, development was unsuccessful but is included here to show how pupation can take place inside the wood.*

Smith, pers. comm.). Usually the cell will be "attached" to one of the sides of the box. If space is not a limiting factor, the head will be directed upwards when pupating. Pupation lasts for 3-4 weeks.

For easy observation of the developmental stages, a few individuals have successfully been reared in petri dished filled with fine substrate material (minimum 9 cm diameter, 1.5 cm height) (fig. 35). This method has been helpful for observing changes in developmental stages, end and resumption of feeding, pupation, and enclosion without disturbing all larvae and pupae by invasively searching through all the substrate in the boxes.

***Figure 35**. Petri dish with substrate and larva. The low height of the dish makes observation of the animal easy.*

2.4.5 Population management

The species is bred at CPH ZOO only for conservation and population management purposes and is therefore (as well as due to the adults' short longevity) not exhibited. The species is bred at the zoo's breeding facilities and larvae are released into artificial nest boxes as well as in natural cavities in the project area each fall. By fall 2020, close to

2500 larvae had been released in Denmark. The captive population consists of 81.81.40 larvae by October 2020.
No other institutions currently hold the species. There is no EEP for the species.

As the species will easily produce large numbers of offspring during each generation, the population might grow very big relatively fast. CPH ZOO plans to continue reintroducing a number of larvae to the wild in the following years, and the captive population size will be controlled accordingly. Euthanatizing should follow the "EAZA Guidelines for the Euthanasia of Invertebrates 2014", but culling can also be done by physical crushing or freezing of larvae.

2.5 BEHAVIOURAL ENRICHMENT

No direct behavioural enrichment seems to be needed for the species. Providing a naturalistic captive environment (with e.g. branches to climb, substrate to bury in, wood lumps to chew galleries in etc.) give the beetles and larvae the opportunity to express a range of natural behaviours.

2.6 HANDLING

2.6.1 Individual identification and sexing

Individual identification is not possible unless beetles are marked. This can be done by gluing (super glue works perfectly) numbered plastic discs (which are usually used to mark queen bees by beekeepers) to the protonum or elytra of the beetles (fig. 36). The glue should be allowed at least 5 minutes to completely dry before releasing the beetle

into its enclosure (or in the field), as the plate might be "pushed" off the protonum and either fall off or end up being stuck to other unintended body parts.

***Figure 36**. Adult marked with numbered plastic discs glued on to the protonum.*

Imagoes can also be marked using permanent markers or color used for marking of bee queens. Other marking techniques include puncturing or drilling holes in the elytra; however, this has not been tested in *G. nobilis*. For an overview of different marking techniques for insects, see Hagler & Jackson (2001) and Goldwasser, Schatz, & Young (1993) for Scarabaeidae specifically.

Sexing of larvae can be done from the third larval stage by looking at the ventral side of the posterior segment of the larva's body. The males have the so-called "Herold's organ" while the females do not (fig. 10 and 11). The presence or absence of Herold's organ can be seen when the larva has molted into L3 but it becomes more visible a few weeks after molting. Sexing of adult beetles is likewise relatively easy. The females' pygidium is notched while the males' are not. Furthermore, the second segment of the second leg pair (mesotibia) of the males are broader than the females' (fig. 15). No significant sexual size or weight differences has been found for neither larvae nor beetles bred at CPH ZOO (and field data is not currently available).

If the animals are left to breed on their own (i.e. without intervention or moving of individuals between boxes) the total number of eclosed imagoes in a box can be difficult

to count directly as the beetles will use a proportion of the time under the substrate surface. The number can however be determined long after the imagoes have died. Many parts of their exoskeleton will remain intact (but not cohesive to the whole body of the beetle) after months laying in the substrate. Especially the protonum (see fig. 14) is a good indicator for the number of imagoes, as its hard, outer shell remains intact and because each beetle only has one single feature of that body part (compared to e.g. the legs or elytra). The protonum is easy to find in the substrate due to its size (compared to the smaller head for example) and due to the shining green color (compared to the ventral parts of the exoskeleton which are primarily black). Thus, one protonum in the substrate will account for one adult beetle (fig. 37). However, the protonum cannot be used to identify the sex of those specific beetles. If the sex is important to know, it is recommended to search for dead imagoes soon after they have died so whole bodies are found. If waiting too long to search, the bodies will most likely be decomposed in the substrate (and according to Whitehead (2002), larvae in the substrate may also eat the females who have buried themselves in the substrate after laying eggs and dying).

***Figure 37**. Several protonum from dead adults found in the substrate of a breeding box. Here, at least 13 adults had eclosed.*

2.6.2 General Handling

Though handling of especially adult beetles does not seem to have any harmful effects on them, it is recommended to minimize handling of all life stages. Handling of pupae should be completely avoided as this might disturb the development and lead to death. If larvae need to be counted or moved from the box in which they hatched, it is recommended to have the females lay her eggs in substrate without any big wood lumps. If left in substrate with big wood lumps the larvae will most likely chew their way inside these and may then be difficult to find. Alternatively, eggs or newly hatched larvae should be moved before hiding inside the wood lumps.

2.6.3 Catching/Restraining

The delicate eggs and small larvae can be handled by using a fine paint brush or, extremely carefully, with tweezers or by fingers. Larger larvae should also be handled gently but can easily be picked up by fingers. Larvae might “attack” when handled by trying to bite, however the bite is not very painful, not even by the third instars whose mandibles seem quite strong. The easiest way to avoid being bitten is to hold on each side of the larvae between the thumb and index finger. When sexing the larvae, the larvae can be held like this as well for an easy visual inspection of the presence of the Herold’s organ.

Wearing disposable gloves will minimize the pain if bitten (and prevent a potential spread of fungal diseases although this has not yet been observed), but please note that wearing gloves might inhibit safe manipulation when handling eggs and small larvae by fingers.

Beetles are easy to handle and will easily walk on human hands. Beetles can be gently picked up by fingers on either side of the elytra or, placing a hand in front of them, they will crawl up onto it by themselves. Feet will hook quite strongly to branches etc., so care should be taken when lifting the animals to not damage their limbs. The beetles can be relatively fast in take-off and flight; however, captive beetles do not seem to attempt escape by flying very often.

2.6.4 Transportation

For brief transport, ensure that the box has air holes and some substrate (preferably similar to the substrate used in the breeding boxes) and that the lid is secured. If transporting adult beetles, add some material that the beetles can cling onto (e.g. some branches). Larvae nearing pupation as well as pupae should not be transported to avoid disturbance.

If transporting the animals by air or courier, the relevant legislation of the newest IATA ‘Live Animal Regulations’ (IATA, 2021) should be followed.

2.6.5 Safety

Make sure to avoid escape of beetles when e.g. examining the breeding boxes, especially if kept outdoors. Beetles might be nearly impossible to locate once escaped and the introduction of the species to areas in which it is not naturally found is of course not desired. However, the species constitute no safety hazards to humans or other animals.

2.7 VETERINARY: CONSIDERATIONS FOR HEALTH AND WELFARE

See section 2.8.

2.8 SPECIFIC PROBLEMS

From other beetle species developing in soil (Sun beetles, *Pachnoda marginata*, and Hercules beetles, *Dynastes hercules*) it has been observed that fungal mycelium (of unknown species) can enter the pupal cases and kill the pupae. Though this has not been directly observed in *G. nobilis* pupae, special attention on this matter is recommended. Larvae and adults are not known to be negatively affected by fungal mycelium.

It is important to ensure that no unwanted species (such as millipedes, centipedes, and other beetle) are present in the substrate before being used due to the risk of predation on especially eggs and small larvae. This is done by freezing the substrate for at least 24 hours (but preferably up to one week) before use. Furthermore, infectious fungus and diseases are avoided by not using any chemicals for cleaning of the boxed used for *G. nobilis* in any life stage. The boxes can be cleaned in the dishwasher at min. 92°C.

The CPH ZOO population is founded only by 1.2 wild-caught beetles. After captive breeding over four generations, no signs of negative effects due to inbreeding have been observed, however this should be kept in mind as a potential future problem. Owen (1993) observed malformed elytra on some adults (in a group founded by one female) and suggested that that could be a result of inbreeding (or potential overcrowding), but this is the only report on potential problems with inbreeding in the species. According to Matthew Smith (pers. comm.), saproxylic beetles with limited dispersal abilities will probably not have a problem in this matter. The group, established by only a single female by Owen (1989), persisted for more than 25 years which indicates that it is not vitally

important that fresh genes enter the population rapidly. In wild populations, the species should be able to wait a long time for another rare occurrence of "limited dispersal" to happen to bring new genes into the population. In *Osmoderma eremita*, another species of tribus Trichiini, it has been found that only approx. 15% of the adults leave their natal site, suggesting that inbreeding is not a big problem here. As *G. nobilis* shows many similar ecological traits as *O. eremita*, this could also apply to our species of interest.

It is anyways recommended that new genes are added to the gene pool on an ongoing basis if possible as the dispersal abilities of the species and the long-term effects of potential inbreeding are not understood in depth and therefore need further investigations.

2.9 RECOMMENDED RESEARCH

There is a great lack on information on *G. nobilis*. Further research could therefore be extremely useful for future conservation planning and management of the species. At CPH ZOO, the lifecycle is currently being studied to determine the length of each developmental stage with comparisons of the indoor and outdoor group (i.e. laboratory conditions vs. more natural like conditions). Furthermore, studies on the substrates' effect on growth and development of the larvae are planned to be carried out in the nearest future.

Other potential areas for further research include optimal densities for captive breeding and investigation of potential negative effects of inbreeding. Furthermore, more detailed field studies are needed, including investigations of growth and development in the natural environment, the ecological role of *G. nobilis* in its microhabitat, dispersal abilities, among others.

Section 3

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