Nutsedge

Biology and Control of *Cyperus rotundus* and *Cyperus esculentus*, review of a literature survey

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1. Introduction

In the framework of the Montreal Protocol international agreements between countries were made about phasing out ozone depleting substances. In 1992 methyl bromide was added as controlled substance to the Montreal Protocol. Since January 2005 the use of methyl bromide for developed countries is prohibited. Critical use exemptions (CUE's) may be requested for specific applications only. These requests are reviewed under the Montreal Protocol and provided with recommendations by the Technical Option Committee for methyl bromide (MBTOC) before parties proceed to decision-making. Decision Ex.I/4 of the Protocol requires Parties which have CUEs and Parties which no longer consume MB to submit information about available alternatives for a database on methyl bromide alternatives. In this country methyl bromide was completely phased out for soil disinfestation already by 1992 due to environmental and public health aspects (Goud 2004). For that reason many alternatives to methyl bromide were tested in the last two decades. These research data however are not easily accessible for anyone, not in the least because many publications are written in the Dutch language.
The Ministry of Housing, Spatial Planning and the Environment (VROM) from the Netherlands has decided to support this data base with Dutch research data in order to stimulate the practical application of alternatives to methyl bromide in countries with comparable problems.
VROM has asked Wageningen University and Research Centre to produce three reports with expert knowledge about important items nowadays: production of strawberry runners, low-cost soilless systems and *Cyperus* spp. weed control. The perennial weeds *Cyperus esculentus*, yellow nutsedge and *Cyperus rotundus*, purple nutsedge are difficult to control without methyl bromide. Yellow nutsedge is a problem in the temperate areas of the world, and purple nutsedge is a notorious weed in tropical regions.
This report presents a review of of the available knowledge on the geographical distribution, the life cycle, non-chemical management options such as crop choice, nutrient management, mechanical operations, available chemical and biological control of *Cyperus rotundus* (purple nutsedge) and *Cyperus esculentus* (yellow nutsedge) based on Dutch literature as well as on foreign literature.

2. Nutsedge; biology

The family *Cyperacea* includes approximately 3000 species of which about 220 species are identified as weeds and of which 42% of these weeds are in the genus *Cyperus* (Bendixen and Nandihalli 1987).
Both purple nutsedge (*Cyperus rotundus* L.) and yellow nutsedge (*C. esculentus* L.) are problem weeds in many parts of the world (Wills 1987). Usually yellow nutsedge is found on low, moist areas, while purple nutsedge is found on well-drained soils (Wills 1987). In mixed stands, purple nutsedge is distinguished by its red, reddish-brown, or purplish-brown loosely arranged inflorescence, dark green leaves which grow low to the ground with boat-shaped leaf tips, and scaly rhizomes which when mature become wiry and hard to break, and produce tubers and bulbs in chains (Wills and Briscoe 1970). Yellow nutsedge has a yellowish-brown or straw-coloured inflorescence which is arranged along an elongated axis in the shape of a bottle brush. It has pale green leaves which grow upright with long needle-shaped leaf tips and weak, easy-to-break rhizomes which often end in bulbs or single tubers but rarely form chains of tubers (Wills 1987).

2.1 Geographic Distribution

The Netherlands
Cyperus esculentus, yellow nutsedge, is the most widespread nutsedge species in the Netherlands. Purple nutsedge has never been observed in the Netherlands. *C. esculentus* was introduced in the seventies of the twentieth century (Lotz, Groeneveld et al. 1991), probably from the USA as a contaminant of shipments of gladiolus cormlets (Groenendael and Habekotté 1988). Given the genetic variation of this material, the introduction must have taken place on several different occasions. The gladiolus was grown on new soils each year to reduce the risk of soilborne diseases. This resulted in a quick spread of yellow nutsedge in a relatively large part of the country (Rotteveel and Naber 1988). In the Netherlands the species is found in crop rotations such as potato-sugarbeet-winter-wheat, tree nurseries, bulb growing, maize-sugarbeet-potato and potato-sugarbeet-onion-winter-wheat-winter-barley (Rotteveel and Naber 1988).

World wide
Nutsedges originate from tropical and subtropical areas, but the tubers are frost tolerant up to a certain degree (Groenendael and Habekotté 1988) and as a result the species can establish itself in more temperate parts of the world as well. In Europe, the species is dominant in South-Europe, but is encountered nowadays in North Western Europe in Germany, Switzerland, France, Austria, The Netherlands and Belgium as well (Naber and Rotteveel 1986; Schippers, Ter Borg et al. 1995). In the USA the species can be found in all states where cotton is grown, such as Arizona, California, New Mexico, Alabama, Arkansas, Florida, Georgia, Louisiana, Mississippi, Missouri, North Carolina, Oklahoma, South Carolina, Tennessee and Texas (Keeley and Thullen 1993). The species can also be found in the North and Middle of the American continent in Canada, Alaska, Cuba, Nicaragua, Puerto Rico and Mexico (Bendixen and Nandihalli 1987).
On the South American continent, nutsedges are present in Peru, Chili, Argentina, Colombia and Venezuela (Bendixen and Nandihalli 1987). In Asia it has been reported in Japan (Li, Shibuya et al. 2004), Indonesia, Taiwan (Bendixen and Nandihalli 1987) and India (Schippers, Ter Borg et al. 1995). *Cyperus esculentus* can be observed frequently on the African continent (Schippers, Ter Borg et al. 1995): Angola, Kenya, Madagascar, Mozambique, South-Africa, Tanzania, Swaziland, Cameroon, Ethiopia, Ghana, Ivory Coast, Mali, Mauritania, Nigeria en Senegal (Bendixen and Nandihalli 1987). In fact, nutsedge can be found on all continents, except for Antarctica (Bendixen and Nandihalli 1987), and is a weed in 21 crops in 40 countries and considered a problem weed in 15 countries (Bendixen and Nandihalli 1987). The main crops in which nutsedge infestation was found were: cotton, maize, rice, cereals, coffee, peanut, pineapple, potato, soya, sugarbeet, and

several vegetable crops (Bendixen and Nandihalli 1987). The distribution of yellow and purple nutsedge appears to be limited by the environment (temperature range and moisture level) rather than the means of dispersal (Bendixen and Nandihalli 1987). Purple nutsedge is limited to areas in which the average minimum air temperature is higher than -1 °C (Bendixen and Nandihalli 1987).

2.2 Life cycle

The life cycles of yellow and purple nutsedge contain many similarities and show some differences between the species. Both species show vegetative activity which produces a complex underground system of basal bulbs, rhizomes and tubers. The basal bulbs form the starting point for the vegetative growth, because they contain the meristems for leaves, rhizomes, roots and flower stalks. The tubers contain quiescent buds and function like the seeds of annuals, acting as the primary dispersal units (Stoller and Sweet 1987).

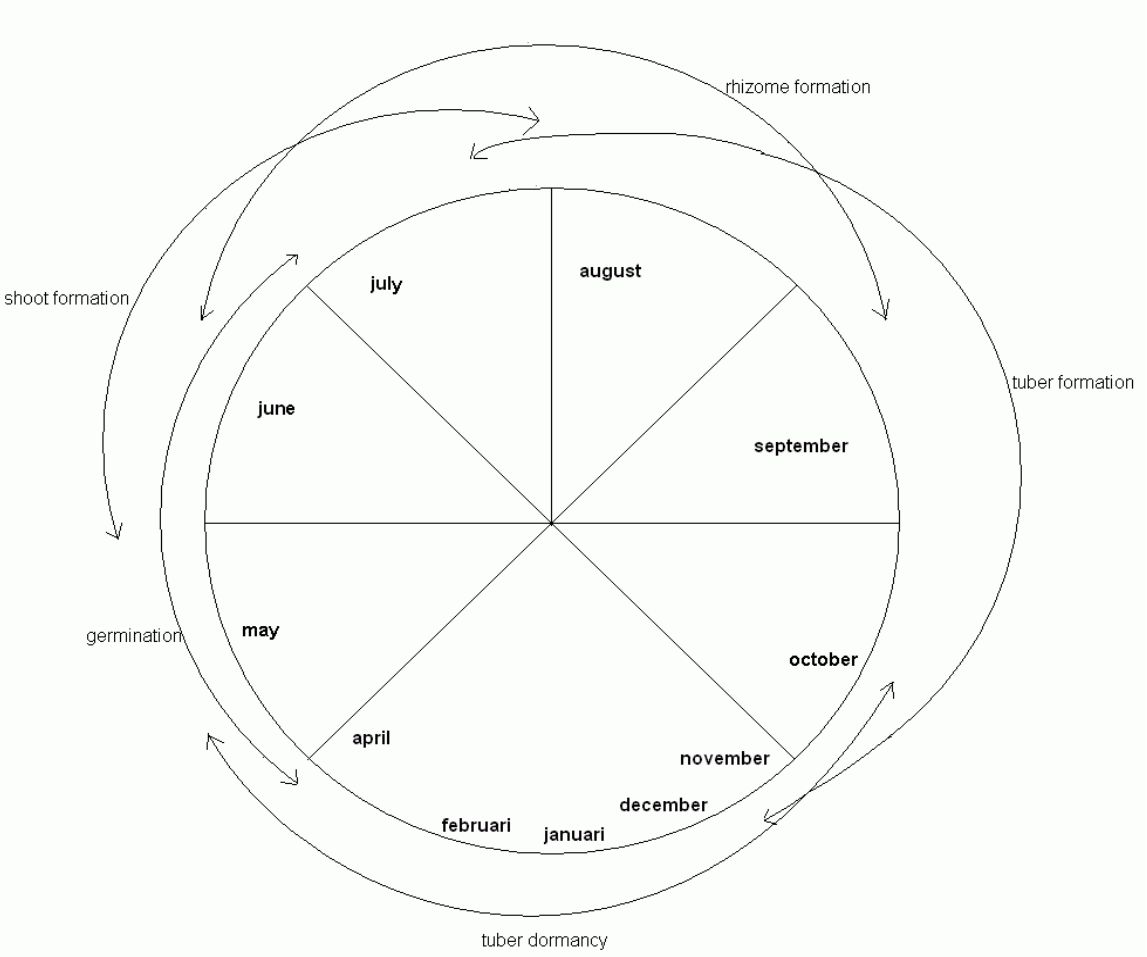

Figure 1 Lifecycle of Cyperus esculentus under Dutch conditions, after Groenendael and Habekotté, 1988.

2.2.1 Reproduction

Reproduction through seed

Yellow nutsedge reproduction through seeds has not been observed in the Netherlands (Groenendael and Habekotté 1988; Lotz, Groeneveld et al. 1991) and is of minor importance in temperate areas of the world (Naber and Rotteveel 1986). Purple nutsedge is a perennial which

rarely reproduces through seeds (Thullen and Keeley 1979). The minor importance of reproduction via seeds is confirmed by the observation that a population present in one field is often the offspring of one or only a few plants (Horak and Holt 1986).

Yellow nutsedge flowers at day lengths from 12 to 14 hours (Jansen 1971). The average number of flowers per inflorescence of *C. esculentus* under field conditions in California varied from 1227 to 6685. The highest percentage of flowers with seeds per inflorescence found in that area was 17% (Thullen and Keeley 1987). The germination percentage of those seeds in climate chambers was on average 31% (Thullen and Keeley 1987).
Germination percentage increases with seed size; the larger the seed, the larger the chance that it will germinate. Seeds of 0.15 mg or less are not capable of germination (Thullen and Keeley 1987). The germination percentage is positively related with temperature. At increasing temperature, an increasing germination percentage can be observed with an optimum at 27/21 °C (Thullen and Keeley 1987). Germination in complete darkness was less than germination under a regime of 11 hours light followed by 13 hours of darkness (Thullen and Keeley 1987).

Vegetative reproduction

Tubers are recognized as the primary dispersal unit for both species (Stoller and Sweet 1987).
In spring, the plant sprouts from tubers in the soil (Groenendael and Habekotté 1988; Defelice 2002) as soon as the soil temperatures reaches a minimum of 8 to 10 °C for yellow nutsedge (Groenendael and Habekotté 1988) and a minimum of 20°C for purple nutsedge (Horowitz 1972). This may partly explain the difference in climatic distribution between the two species; yellow nutsedge is distributed in colder regions of the world than purple nutsedge.
Yellow nutsedge buds cluster at the apical end of tubers (Bendixen 1972), while purple nutsedge buds cluster at the nodes along the entire end of the tubers.
The sprouting bud forms one to two upwardly growing rhizomes that produce the basal bulb under the influence of light (Groenendael and Habekotté 1988). In purple nutsedge basal bulbs are similar to tubers in appearance and sprouting characteristics (Stoller and Sweet 1987). From this basal bulb the aboveground plant emerges in a typical sedge like manner. The shoots can reach a height of 10 to 90 cm (Defelice 2002). After a few weeks lateral rhizomes will grow from this basal bulb that will grow and elongate up to 1 m (Groenendael and Habekotté 1988). The lateral rhizomes will give rise to the formation of secondary sprouts that will form tertiary sprouts, and so on. The formation of the lateral rhizomes from the basal bulb starts 4 to 6 weeks after the appearance of the first aboveground shoots (Stoller and Sweet 1987). More than 95% of purple and yellow nutsedge tubers usually are formed in the upper 45 cm of the soil (Stoller and Sweet 1987) and in most soils more than 80% of the tubers occur in the upper 15 cm. Rhizomes do not penetrate deeply in heavy soils, so tubers are distributed deepest in light-textured soils (Stoller and Sweet 1987).
From July more and more rhizomes will grow downward and produce new tubers instead of shoots. In yellow nutsedge, this switch from sprouting rhizomes to tuber forming rhizomes is influenced by photo-period (Stoller and Sweet 1987; Groenendael and Habekotté 1988), N-availability, density and light penetration, nitrogen availability, density and light penetration of the soil (Groenendael and Habekotté 1988). Short photoperiods stimulate reproductive growth, long photoperiods stimulate formation of basal bulbs and shoot formation (Stoller and Sweet 1987).

Light intensity also strongly influences the number of tubers per plant produced and the individual weight per tuber formed. At high light intensities (37, 5 Wm^{-2}) in greenhouse experiments, tubers produced more than 500 tubers per plant, three hundred tubers were produced at intermediate light intensities (16,3 Wm^{-2}) and less than fifty tubers at low light intensities (6,9 Wm^{-2}) (Lotz, Groeneveld et al. 1991). The weight per tuber also decrease with the light intensity; from 92 mg per tuber at the highest intensity, to 52 mg and 27 mg at the intermediate and lowest intensities, respectively (Bärring 1986).

Tuber production in the field can be very large; in temperate areas up to hundreds per plant and in tropical areas up to thousands (Naber and Rotteveel 1986). When purple and yellow nutsedge are cultured in fields without interference from other plants, they can produce 10 to 30 million tubers per ha in a season (Hauser 1962; Horowitz 1972).

The number of tubers per basal bulb that can be produced varies with the length of the growing season as well (Thullen and Keeley 1987). The longer the growing season, the larger the number of tubers being produced (figure 2 right). On top of that, this number also varies with the moment at which the basal bulb is planted (figure 2 left).

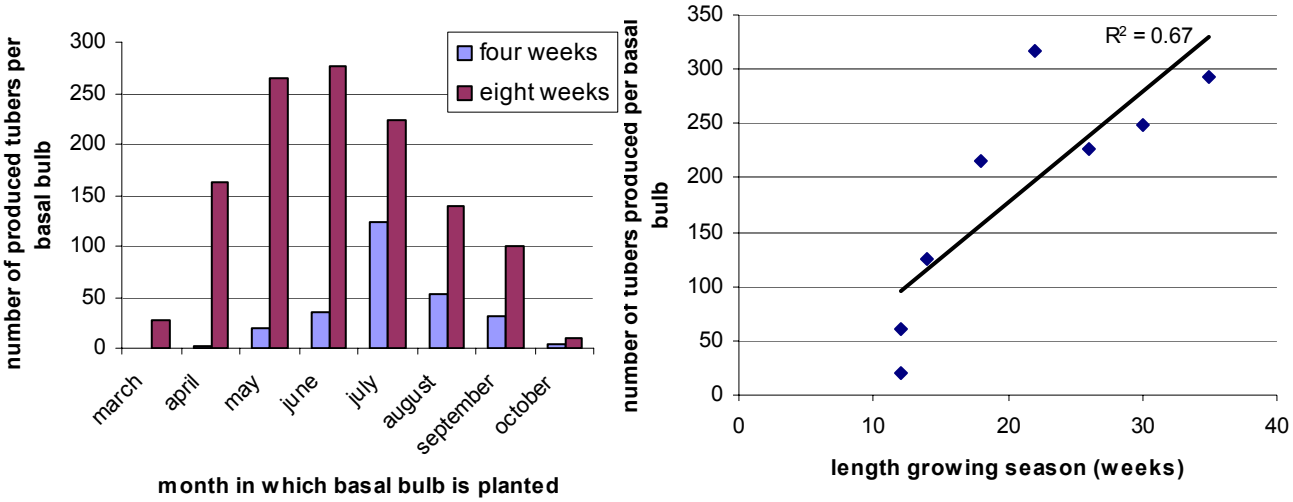

Figure 2 Influence of the length of the growing season on the number of tubers being produced. Left: effect of planting moment of the basal bulb on the number of produced tubers in a growing season with a length of 4 or 8 weeks. Right: the effect of the length of a growing season with a length of 12 weeks or more on the number of produced tubers. After Thullen and Keeley, 1987.

In optimal (sub)tropical conditions, a tuber can produce up to 17000 tubers in one growing season, but in temperate zones a tuber produces on average 500-600 tubers (Groenendael and Habekotté 1988).
Day length does not affect tuberization dramatically in purple nutsedge as it does in yellow nutsedge (Williams 1983). Purple nutsedge tuberization can occur all year in tropical climates and may be a response to excess carbohydrate, regulated by temperature and possibly day length (Hammerton 1975). Horowitz (Horowitz 1972) did not find any effects of natural photoperiods on tuberization of purple nutsedge, while Hammerton (1975) showed that day length was the main factor driving purple nutsedge growth and development (Hammerton 1975).

In autumn the aboveground parts and the rhizomes directly connected to these parts die. At that moment new tubers can now germinate provided that the temperature is high enough and tubers are nondormant. The dormancy of these tubers increases with increasing burial depth (Groenendael and Habekotté 1988).
See also figure 1 for a schematic overview of the life cycle of *C. esculentus* in temperate regions. Most new tubers (80%) are formed in the top 15 cm of the soil. They are capable however of growing up to a depth of 40 cm (Groenendael and Habekotté 1988).

2.2.2 Dormancy

Yellow nutsedge tubers are most dormant at the end of the growing season in which they were formed, and less dormant at the beginning of the next growing season in spring (Taylorson 1967).

In temperate climates, cold winter temperatures and leaching with water are factors that promote germination. Besides all chemical and physical factors such as ethylene, O_2, and gibberellic acid and desiccation and scarification, tillage operations can break dormancy as well (Taylorson 1967). Soil warming is considered the major sprouting stimulus in temperate climates, while soil moistening is a sprouting stimulus in arid climates (Horowitz 1972).

2.2.3 Ecotypes

Cyperus esculentus has a high phenotypical plasticity (Schippers, Ter Borg et al. 1995), which makes the species difficult to identify and at the same time difficult to control as well. Significant differences between ecotypes (populations collected at different locations) in rhizome and tuber development, flowering and responses to herbicides (Stoller and Sweet 1987), emergence period, first flowering, plant height, aboveground biomass, rhizome length, size of the petioles and seeds (Holt 1994) exist.

Reports of ecotypic variation in several important biological characteristics such as tuber dormancy and longevity (see also paragraph dormancy), exist (Stoller and Sweet 1987).

Japanese clones of *C. esculentus* can regulate their way of reproduction under the influence of water. The plants produce relatively more tubers (vegetative reproduction) in very humid conditions, while they produce relatively more seeds under dry conditions (Li, Shibuya et al. 2001). In general, the species produces more shoots and tubers under humid than under dry conditions (Li, Shibuya et al. 2001). Differences in response to environmental factors may reflect local ecotypic adaptation to several environmental conditions including photoperiod, temperature, and moisture conditions (Stoller and Sweet 1987).

2.2.4 Leaf formation

The development of the leaf vascular system appears to be similar for both purple and yellow nutsedge (Wills, Hoagland et al. 1980). The leaves grown from the bulb in an unfolded triangular fascicle whose development on the bulb begins at the outermost leaf, progresses inward and terminates with a seed-bearing rachis. The vascular system of an individual leaf begins with three main bundles originating as the extension of three vascular strands into the leaf primordium. Additional vascular bundles are formed by divisions of the original vascular strands. The development of all new leaf tissue, including the vascular system, occurs mainly in the intercalary meristem at the base of the leaves. Translocation of a herbicide from leaves into the rest of the plant may be restricted by the disconnected accessory bundles and/or by the immature vascular elements (Wills 1987).

2.2.6 Survival

Due to apical dominance and bud dormancy, tubers stay in the soil for extended periods before sprouting. Control would be facilitated if tuber longevity were short enough so that all buds could sprout at the same time so that the resultant plants can be killed (Stoller and Sweet 1987).

Yellow nutsedge tubers were found to have a half life of 4 and 6 months in a non-crop environment in the US at depths of 10 and 20 cm, respectively (Stoller and Max 1973). In a continuous maize crop which was moderately infested with yellow nutsedge, a minimum of 2 years season long chemical control was required to reduce the tuber populations with 80%, and another year of season long chemical control was needed to reduce the population to 95% of the original population (Stoller, Wax et al. 1979).

It is known that desiccation and extremely low temperatures can kill the tubers of both yellow and purple nutsedge (Thomas 1969; Stoller and Sweet 1987).

When purple nutsedge tubers are dried out until their water contents is 15% (normally they contain 85% water), they will not survive. However, it may take several days or even weeks to achieve this and is therefore highly weather dependent (Stoller and Sweet 1987). Effects of desiccation on yellow nutsedge tubers are less clear and seem to vary with ecotype (Thomas 1969). In general, yellow nutsedge tubers are capable of tolerating drying better than purple nutsedge tubers under both field as well greenhouse conditions (Bendixen and Nandihalli 1987).

Yellow Nutsedge survives the winter through the formation of tens of small tubers which are formed at the end of rhizomes during summer (Naber and Rotteveel 1986). Tubers are capable of surviving certain periods of frost. Yellow nutsedge can survive lower temperatures than purple nutsedge. In a lab study with ecotypes from the US, yellow nutsedge tubers were killed by 50% at a temperature of -6.5 °C and purple nutsedge tubers at a temperature of -2 °C at an exposure time of four hours or longer (Stoller 1973).
In a Dutch study, tubers of yellow nutsedge were able to withstand this temperature for a longer period of time. Table 1 shows the percentage of yellow nutsedge tubers, that was still capable to grow after a certain period of frost (Groenendael and Habekotté 1988). Differences in the hardiness are probably linked with the ecotypic variation.
Because of the relatively mild current Dutch winters the viability of the tubers will not strongly decrease as a result of low temperatures. Survival of tubers is the largest in deeper soil layers (Stoller and Sweet 1987).

Table 1 Survival of large (0.174 gram) and small (0.048 gram)C. esculentus tubers during different frost periods (Groenendael and Habekotté 1988).

Temperature (°C)	0			-2			-4		
length of period (days)	2	8	32	2	8	32	2	8	32
% emergence (large tubers)	62	62	43	50	43	37	43	62	0
% emergence (small tubers)	58	43	65	58	14	7	50	36	7

3. Management

3.1 Non-chemical

Several papers and reports present the results of experiments investigating the possibilities for non-chemical nutsedge control. In the following paragraphs, an overview of the most relevant and important results is described.

3.1.1 Competition and crop choice

Yellow nutsedge is a C4-species and requires a lot of light for optimal growth. Therefore, crops with an early closing canopy restrict nutsedge growth, whilst crops with an open or late closing canopy favour the weed (Rotteveel and Naber 1988).

Field crops
A Dutch experiment with fodder maize, hemp, winter barley, fallow land and winter rye showed that yellow nutsedge was able to produce the most tubers in a fallow field (171,2 per plant) and the lowest in hemp (0,2 per plant) (Lotz, Groeneveld et al. 1991). There was no difference between winter barley and maize in the number of *C. esculentus* tubers produced. The experiment was performed with pots with 3 tubers per pot, which were placed in the soil in the field in such a manner that the surface of the soil in the pot was level with the soil in the field between the crop plants in May. At the end of the season, end October; the dry weight and the number of tubers were determined per pot.
At the same time, the effect of crop type was investigated with naturally occurring *C. esculentus* populations. The naturally occurring population did not show differences in the number of primary rhizomes of nutsedge between crops. However, the number of secondary roots, the aboveground biomass and the number of secondary rhizomes differed significantly between crops. Hemp was able to reduce the *C. esculentus* population the strongest. Compared with fallow land, the growth of maize, winter barley and winter rye reduced the number of aboveground biomass and the number of secondary rhizomes with 39, 29, and 50% respectively. The individual tuber weight did not differ significantly between the crops (Lotz, Groeneveld et al. 1991). Greenhouse replacement series performed by (Collins and Chase 2007) show that cowpea and velvet bean are equally competitive with yellow nutsedge, and that sun hemp, although its height is larger than that of the other two crops, is less competitive. The higher percentage photosynthetically active radiation reaching the soil layer in hemp compared to the other crops may be the reason for this observation. The leaf area per plant of sun hemp was 63 to 70% of cowpea and only 37 to 41% of velvet bean.

Competition experiments (Tuor and Froud Williams 2002) with purple nutsedge- maize mixtures and purple nutsedge- soybean mixtures show that both root and shoot competition (for light and nutrients) from purple nutsedge affect the growth and development of both soybean and maize. Root competition is relatively more important than shoot competition. An increase in yield of purple nutsedge resulted in a decrease in the yield of maize. The species were found to be mutually exclusive. The early establishment and rapid growth rate of purple nutsedge (due to its reserves in the tubers) offers a competitive advantage over maize and other annual crops. Application of additional nitrogen (120 kg N per ha) did not alleviate the competitive stress imposed on both maize and purple nutsedge, although N may be limiting. It is possible that the additional nitrogen stimulated the growth of both, thereby increasing the demand for other nutrients which also may be limiting.
Purple nutsedge yield was unaffected by the presence of soybean, whereas the soybean yield was lower when grown in the presence of purple nutsedge. This may be the result of competition for resources and/or the production of phytotoxins (allelopathy) by purple nutsedge. The addition of

nitrogen did not affect the yield of one of the species, indicating that competition occurred for other nutrients or that the species were not capable of utilizing the resources available (Tuor and Froud Williams 2002).

Purple nutsedge in a cotton crop could be suppressed by intercropping the cotton with single and double rows of sorghum, soybean and sesame (Iqbal and Cheema 2007). The crops were capable of reducing purple nutsedge density with 70-96% and dry matter production with 71-97%. The cotton yield itself was suppressed by the intercrops as well with 8-23%, but this was far less severe than the yield loss caused by the purple nutsedge. On top of that, its loss due to the intercrops was compensated by the greater total economic returns (20% more for sorghum and sesame than cotton alone).

Tomato
The difference between full interference (above- and belowground) and partional interference (only belowground or only aboveground) of yellow and purple nutsedge with tomato was investigated in container experiments in the greenhouse (Morales-Payan, Stall et al. 2003). Full interference by yellow nutsedge was more detrimental to tomato than full interference by purple nutsedge; yellow nutsedge reduced the shoot dry weight of tomato with 34% and purple nutsedge with 28%. Their was no difference in the effect of below- and aboveground interference of yellow nutsedge on the tomato shoot dry weight (both reduced the tomato with 19%). Belowground purple nutsedge interference reduced tomato shoot dry weight stronger (18%) than aboveground interference did (9%). The belowground interference resulted in a nitrate shortage in the sap of the tomato. Purple nutsedge growth was influenced more strongly by tomato shading than by belowground interference from the tomato, whereas yellow nutsedge showed no difference.

Watermelon
In watermelon the critical density in both direct- seeded and transplanted watermelon is 2 yellow nutsedge plants per m^2 to prevent yield losses greater than 10%. Transplanting melons does not improve their competitive ability with yellow nutsedge. At a density of 12 yellow nutsedge plants per m^2 more than 40% yield loss was found for watermelon in both direct seeded and transplanted melons (Buker III, Stall et al. 2003).

Crop residues
The production of yellow nutsedge tubers can be reduced by adding dry (2 weeks at 60 °C) rests (particles smaller than 1 mm) of Raphanus *raphanistrum*. This effect was found in a greenhouse pot experiment with pepper and tomato in which the soil was enriched with 1% *R. raphanistrum* particles. The total biomass of the nutsedge plants remained unaffected when they were grown in the enriched soil in a monoculture. However, when they were grown in a mixture with another species, the tuber weight decreased from 0.32 gram per tuber (without soil enrichment) to 0.05 gram per tuber (with *R. raphanistrum* particles) and was the tuber production reduced by 88%. The competitive ability of yellow nutsedge strongly decreases with the addition of *R. raphanistrum* compared to other crops. The competitive ability of nutsedge is probably strongly dependent on the presence of mycorrhizae. Isothiocyanates that are released by *R. raphanistrum* are toxic for mycorrhizae(Norsworthy and Meehan 2005).

Yellow nutsedge emergence in spring was not hampered by the rests of *Vicia villosa* Roth, when those were incorporated in the preceding autumn (September) (Teasdale and Rosecrance 2003).

3.1.2 Nutrient management

In general, high levels of nutrients are favourable for the number of tubers that is developed, the dry matter level of the rhizomes (g) and the total dry matter content of the plant (Li, Shibuya et al. 2004). However, the relative weight of the rhizomes (per gram dry matter of the plant) and the

relative number of tubers (per gram dry matter of the plant) does not increase with increasing nutrient level (Li, Shibuya et al. 2004).

Nutrient level seems to be important for mowing regime as well. Li (2004) found that reducing the aboveground biomass of the plants with 50% by cutting resulted in a reduced tuber size per unit of plant biomass. However, this treatment did not sort any effect when the nutrient levels were very low. Rhizomes can either produce tubers or basal bulbs and shoots. At low nutrient levels the rhizomes don't produce shoots, or very low numbers, but are still able to produce tubers. At high nutrient levels, the rhizomes produce both tubers and shoots. Nitrogen for instance, is known to stimulate the formation of the basal bulb and thereby the formation of aboveground shoots compared to tuber formation (Stoller and Sweet 1987). The removal of aboveground biomass at high nutrient levels results in an attempt of the plant to replace this lost biomass. As a result, temporarily less reserves are being stored in the tubers. At low nutrient levels, the plant is not investing in aboveground biomass anyway, and as a result removing aboveground biomass will not affect the tuber size (Li, Shibuya et al. 2004).

3.1.3 Mowing

According to (Brecke, Stephenson et al. 2005) the control of shoot biomass of yellow nutsedge by mowing (or herbicide applications) may lead to a depletion in the total tuber number and tuber viability by reducing the carbohydrate reserves. (Summerlin, Coble et al. 2000) found that the length of the rhizomes, the number of tubers and the size of the tubers could be reduced by mowing grass 1 to 3 times per week. The mowing height was very important; mowing at a height of 1.3 cm already affected nutsedges after 6 weeks, while mowing at a height of 3.8 cm affected nutsedge 9 weeks after the first mowing treatment.
Similar results were found for purple nutsedge by (Santos and Morales 1997). In a greenhouse experiment tubers of four fresh weight categories (0.25, 0.50, 0.75 and 1 g per tuber) were planted at a depth of 0.50 cm deep in containers filled with potting medium. Primary shoots were removed for the first time after 6, 12, 18, 24 or 30 days after transplanting. When the first shoot was removed at six days after transplanting, the smaller tubers were unable to regrow and were depleted 30 days after planting (0.25 and 0.50 gram). The larger tubers (0.75 and 1.00 gram) were able to recover. When the first removal took place at 12 days after planting, the smallest tubers (0.25 gram) were depleted 42 days after planting, the other tubers were unaffected. No effect of tuber size was found when first removal was imposed at 18 days after planting or later.
These results imply that tubers, independently of tuber size, are capable of regenerating carbohydrate reserves regardless of subsequent number of removals (Santos and Morales 1997).

3.1.4 Compost

A garden compost experiment resulted in the death of yellow nutsedge rhizomes. Rhizomes were buried at a depth of 30 cm in garden compost during 56 days. The temperature in the centre of the compost was 60C. None of the rhizomes survived the treatment (Daugovish, Downer et al. 2007).

3.1.5 Tillage operations and mechanical control

In general, mechanical control is regarded as little effective and is said to help spreading the plant over fields in stead of reducing the population in a field (Rotteveel 1993).

Tillage operations that bring tubers close to the soil surface may contribute to a reduction of the tuber population. The tubers closest to the soil surface will experience most frost and are at risk of desiccation the most. This may especially be an option for the control of purple nutsedge in arid regions, because it is sensitive to desiccation (Stoller and Sweet 1987). See also paragraph on survival.

Teasdale and Rosecrance (2003) report that tandem disk harrowing after emergence of a maize crop reduced the number of *C. esculentus* L. plants per m^2 with 70 to 87%. They counted the aboveground plants 3 weeks after control (Teasdale and Rosecrance 2003).

Fallow tillage during the summer proved to be efficient in reducing the amount of viable yellow nutsedge tubers in a study by (Johnson 2007). They used a power tiller (Kelley manufacturing) that had a working depth of 7.6 cm and applied it either weekly or monthly. There was no difference between the weekly and the monthly treatments. They started the tillage in early May (Johnson 2007).

3.1.6 Soil solarisation

(Rosskopf, Chellemi et al. 2000) mention a study by (Locascio, Olson et al. 1999) in which soil solarisation was investigated as an alternative to methyl bromide in strawberries. They report that soil solarisation for eight to ten weeks controls nutsedge in strawberry fields at two locations. The experiments were conducted at two commercial farms, one experimental farm and one organic farm, over a two-year period whenever possible. The treatments were soil solarisation, soil solarisation in combination with biosolids compost, and solarisation after deep disking. All treatments resulted in less than one nutsedge plant per m^2 emerging through the plastic. They don't mention whether it concerned purple or yellow nutsedge.

According to (Johnson 2007) summer long solarisation from May or July until October effectively control naturally occurring yellow nutsedge populations in the field. Solarisation beginning in September was not effective, unless it was preceded by monthly or weekly tillage. Tillage was performed with a "power tiller" at a depth of 7.6 cm. Soil temperatures at a depth of 10 cm were on average 30°C, whereas soil temperatures at the same depth under a solarisation mulch (clear colourless polyethylene) were on average 40°C (Johnson 2007). The reduction in the number of viable tubers by the solarisation treatments was not only immediately noticeable, but also significant in the following growing season in a maize crop.

3.2 Biological control

(Phatak, Callaway et al. 1987) has listed all natural enemies that were ever reported as natural enemies of yellow and/or purple nutsedge. More than 130 insect species, 26 fungal species, 10 nematode species and 2 viruses and bacteria were found. Most of the fungal and insect species reported about were pathogenic to *C. rotundus*, the nematodes were found an equal number of times on either yellow or purple nutsedge (Phatak, Callaway et al. 1987). The lists previously reported by Phatak *et al*. (1987) are shown in Appendix I, II and III.
A number of these organisms have been proposed as potential agents for nutsedge control, such as *Aleurocybothus* sp*., Antonia australis, Athesapeuta cyperim Bactra minima minima, B. verutana, B. truculenta, B. venosana, Chorizococcus rostellum, Dercadothrips caespitis, Phenacoccus solani, Rhizoecusi cacticans, Shoenabius* sp*., Sphenophorus phoenicicensi* and *Puccinia canaliculata* (Morales-Payan, Charudattan et al. 2005). The pathogens were never regarded as suitable due to insufficient nutsedge control, or were pests, vectors of diseases or diseases of important crops.
The list of fungi genera associated with purple and/or yellow nutsedge includes *Alternaria, Ascochyta, Balansia, Cercospora, Corynespora, Chaetophoma, Cintractia, Claveceps, Cochliobulus,Curvularia, Dactylaria, Dreshclera, Duosporium, Entyloma, Fusarium, Macrophomina, Marasmius, Phaelotrichoconis, Pythium, Phyllosticta, Phytophthora, Puccinia, Rhizopus, Sclerotinia, Septoria, Tanatephorus,* and *Uredo* (Morales-Payan, Charudattan et al. 2005).

Dactylaria higginsii

The commercialization of the fungus *Dactylaria higginsii* (Luttrell) M.B. Ellis as a control agent of *C. rotundus* and *C. esculentus* is being investigated (Kadir and Charudattan 2000). The isolate is highly pathogenic to both sedges and inoculation of purple nutsedge with conidial suspensions in the lab resulted in significant reductions in shoot number, shoot dry weight as well as tuber dry weight. In field trials, three post emergence applications reduced the purple nutsedge population by more than 90%, including significant reductions in the number of tubers that were produced. In greenhouse studies, the competitive ability of nutsedge was decreased by application of suspension of 10^6 conidia/ml so that tomato yields equal to the weed free control could be achieved (Kadir and Charudattan 2000). However, it has been reported that the secondary infection level is too low to provide sufficient levels of control during the growing season (Kadir and Charudattan 2000).
Another drawback of *D. higginsii* application is the sensitivity of this fungus to other pest control measures such as the application of herbicides like oxyfluorfen, sethoxydim, and diuron. These herbicides were found to reduce the germination of *D. higginsii* (Yandoc and Rosskopf 2006).

Puccinia canaliculata

In the US the rust *Puccinia canaliculata* is known and used to control yellow nutsedge(Phatak, Sumner et al. 1983) (“Dr. BioSedge”(Morales-Payan, Charudattan et al. 2005)). The fungus inhibits nutsedge flowering and new tuber formation. Shoot growth and tuber formation can be reduced with 50-75 % and rust infected nutsedge plants are generally more susceptible to herbicides as well (Scheepens and Hoogerbrugge 1991). The fungus also dehydrates and kills nutsedge plants (Phatak, Sumner et al. 1983). The major problem with this fungus is the difficult large-scale production of spores (it is an obligate parasite), and its low efficacy against some yellow nutsedge biotypes and poor low efficacy against purple nutsedge (Morales-Payan, Charudattan et al. 2005). Scheepens and Hoogerbrugge (1991) (Scheepens and Hoogerbrugge 1991) tested a *P. canaliculata* strain from the US in the Netherlands on several Cyperaceae. *Cyperus esculentus* “leptostachyus” from five locations was susceptible to the rust. From two other locations yellow nutsedge (unknown biotype) was moderately susceptible, whereas plants (biotype leptostachyus) from a third location were resistant to the rust. *C. esculentus* “esculentus” (from one location) and *C. esculentus* “sativus” were resistant as well. However, the rust was not introduced in the Netherlands because of its unexpected wide potential host range (Scheepens and Hoogerbrugge 1991).

It is unlikely that any bioherbicide known today will provide full control of nutsedges. Several species do have the potential of being developed into bioherbicides that may play a role in integrated nutsedge management strategies in both conventional and organic production systems (Morales-Payan, Charudattan et al. 2005).

3.3 Chemical control

A lot of research has been done in the past on herbicide efficacy against nutsedge. However, most and in a lot of cases all herbicides that were tested are not allowed anymore and are not an option to improve nutsedge control any longer. Therefore, results from these studies are not included in this report.

Chemical control is complicated and regarded as too expansive in many crops; little chemicals are known that affect nutsedges and at the same time, don’t affect the crop. As a result a nutsedge population in open summer crops such as beets, gladiolus and asparagus can develop very quickly, without being hampered by mechanical or chemical control (Rotteveel 1993).

Research from (Costa and Appleby 1976) shows that different varieties of *C. esculentus* differ in their response to herbicides. For instance, *C. esculentus* L. *var. esculentus*, is less vulnerable to pre- emergence treatment with atrazine or metribuzin than *C. esculentus* L. var.*leptostachus* Boeck. On the other hand, *C. esculentus* L. *var. esculentus* is more sensitive to post emergence treatment with 2,4-D (Costa and Appleby 1976). *C. esculentus* L. *var. esculentus* is the most abundant variety in Europe and Africa, while *C. esculentus* L. *var. leptostachus* Boeck. is the most abundant variety in North and South America (Schippers, Ter Borg et al. 1995).

Alternative control measures that are mentioned are repeated glyphosate application during several years and the use of metam-natrium (Wijchman 2007). However, dosages that are mentioned are too heigh to comply with current and future regulations. On top of that control with metam-natrium (Monam) is usually not enough to control nutsedge and the additional use of other herbicides may be required (www.ctgb.nl). Bentazon (Basagran) can be effective in combination with a mineral oil in the five leaf stage of a maize crop, in beans, and in onions to control yellow nutsedge. However, nutsedge plants should be smaller than 10 cm and it is recommended to use another herbicide in an earlier stage to weaken the plants (www.ctgb.nl).
In a maize crop some effectivity can be expected from metolachlor (Dual) (Werkgroep Knolcyperus NRLO 1989).
Metazachlor (Butisan) is also effective (although less than methyl bromide) but registration is under discussion for some Cruciferous crops in the Netherlands.

4. Summary/ Conclusions on management options

4.1 General information

An infestation with yellow nutsedge will cause serious economic losses to farmers in the Netherlands. On infested fields it is not allowed to grow crops for a period of at least three years. After three years the field can be used again, provided that no nutsedge tubers can be found on the field any more.

The last decade, the problem with yellow nutsedge seems to increase in the Netherlands according to several sources (Knippels 2005; Dwarswaard 2006; Waterink 2006; Wijchman 2007). In 2006 38 new fields were infested with *C. esculentus*, in spite of the strict sanitary regulations in the country (Wijchman 2007). In most of the newly infested fields, Lily was grown (Knippels 2005; Dwarswaard 2006). In almost all cases it concerned bulb crops.
The increase in the yellow nutsedge spread in the second half of the nineties and at the beginning of the 21st century in the Netherlands is said to be the result of a decrease in the alertness of the farmers and landlords, the difficult determination of the species, but also of a denial of the problem when one runs into a nutsedge infestation (Knippels 2005).

4.2 Non-chemical control

Some non-chemical methods such as crop choice can help manage a nutsedge infestation in a field. Crops like hemp, velvetbean, cowpea, sorghum, sesame and Raphanus raphanistrum were found to be capable of competing with nutsedges and in some cases even reduce the nutsedge population in the field. Other crops such as maize, tomato and watermelon need to be avoided when no effective herbicides are available. Glyfosate resistant maize (GMO) with regular glyfosate treatments can be an effective option.
Results on soybean are contradictorily; some sources report of a good competitive ability against nutsedge, while others observed an increase in the nutsedge populations.
In general, crops that close their canopy early and have afast early growth can help manage nutsedge (nutsedge is a C4 species).

Aboveground removal of biomass, for example through mowing, can be effective. At this moment no clear advice about the frequency can be given. Reported frequencies vary from 1 to 3 times a week to every 18 days. Mowing is thought to be effective at high nutrient levels only. Nutsedges don’t form shoots at low nutrient levels and will not use resources from their tubers to replace removed aboveground parts at these levels. At high nutrient levels, the plants will use their belowground reserves to replace aboveground parts.

Tillage operations can be used to control nutsedges as long as they cause the rhizomes and tubers to dessicate, and are applied repeatedly during summer. It is required to fallow the land in those cases. In general, the risk of spreading the tubers over the field by mechanical control and tillage operations is regarded as too large compared to the results that can be obtained.

Results with soil solarisation show some perspective for regions in which soil temperatures of 40 °C can be achieved. Solarisation starting in July was found to be effective, which has the benefit that an early crop can still be grown, or that some tillage operations can be applied to weaken the nutsedges beforehand.

In the Netherlands good results were obtained with covering infested soil with black polyethylene film during a complete growing season as well (Werkgroep Knolcyperus NRLO 1989).

4.3 Biological control

It is not likely that any of the known bioherbicides can give full control of nutsedges on their own. However, some biocontrol agents such as *Dactylaria higginssi* and some *Puccinia* spp. may be included in integrated management systems, provided that other control measures do not interfere with their pathogenicity, conditions are humid enough and the risk of affecting non-target plants such as beneficial wild plant species and important crops is low.

4.4 Chemical control

Chemical control measures that are mentioned are repeated glyphosate application during several years and the use of metam-natrium (Wijchman 2007). However, dosages that are mentioned are too high to comply with current and future regulations. The option of growing GMO Maize or other glyfosate resistant crops makes repeated dosages possible, however, growing GMO maize or other glyfosate resistant crops is not allowed in the Netherlands and most of Europe yet.
On top of that control with metam-natrium (Monam) is usually not effective enough to control nutsedge and the additional use of other herbicides may be required. Bentazon (Basagran) can be effective in combination with a mineral oil in the five leaf stage of a maize crop, in beans, and in onions to control yellow nutsedge. However, nutsedge plants should be smaller than 10 cm and it is recommended to use another herbicide in an earlier stage to weaken the plants.
Metolachlor and metazachlor also have the potential to reduce nutsedge populations.

At this moment, research considering management systems that take into account crop choice, nutrient levels and possible mechanical and chemical control methods is lacking and needed to provide an efficient management system for nutsedges.

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Appendix I.

Natural enemies of *C. esculentus* and *C. rotundus:* insects

Table. Insects feeding on *C. esculentus* and *C. rotundus*. After (Phatak, Callaway et al. 1987)

Organism	Species infected
Aleurocybotus sp.	*C.rotundus*
A. occiduus sp. N.	*C.rotundus and C.esculentus*
Althaes hibisci	*C.esculentus*
Amsacta moorei	*C.rotundus*
Anacentrinus blanditus	*C.rotundus*
Anthomyze sp	*C.esculentus*
Antonina australis	*C.rotundus*
Apis indica	*C.rotundus*
Athesapeuta cyperi	*C.rotundus and C.esculentus*
Bactra bactrana	*C.rotundus*
B. furfurana	*C.rotundus and C.esculentus*
B. lanceolana	*C.rotundus and C.esculentus*
B. minima minima	*C.rotundus*
B. phaeopis	*C.rotundus and C.esculentus*
B. truculenta	*C.rotundus and C.esculentus*
B. venosana	*C.rotundus*
B. verutana	*C.rotundus and C.esculentus*
Barinus squamolineatus	*C.esculentus*
B. curticollis	*C.esculentus*
Barilepis grisea	*C.esculentus*
Calendra sp	*C.esculentus*
Calligypona striatella	*C.rotundus*
Carolinaia cyperi	*C.esculentus*
Chaetocnema pulicaria	*C.rotundus*
Chaetopsis fulvifrons	*C.esculentus*
Chiloides copidotis	*C.rotundus*
Chlorops sp.	*C.esculentus*
Chorizococcus rostellum	*C.rotundus and C.esculentus*
Cisseps fulvicollis	*C.rotundus*
Corimelaena pulicaria	*C.esculentus*
Culex pipiens quinque	*C.rotundus*
Cydia perfricta	*C.rotundus*
Delphacodes puella	*C.rotundus*
Delphacodes basivitta	*C.esculentus*
Deltocephalus sonorus	*C.rotundus*
Diabrotica undecempunctata howardi	*C.esculentus*
Dorcadothrips coespitis	*C.rotundus*
Draculacephala portola	*C.rotundus*
Elachiptera nigriceps	*C.esculentus*
Elasmopalpus lignosellus	*C.rotundus*
Elliponeura debilis	*C.esculentus*
Euscyrtus concinnus	*C.rotundus*
Evylaeus sp	*C.rotundus*
Exitianus exitiosus	*C.rotundus*

Ferrisia virgata	*C.rotundus*
Frankliniella fusca	*C.rotundus*
Gastrimargus transversus	*C.rotundus*
Glyphipterix impigritella	*C.rotundus and C.esculentus*
Glyphipterix prob. Imprigtella	*C.esculentus*
Graminella nigrifrons	*C.rotundus*
Halticus bracteatus	*C.esculentus*
Halictus bracteatus	*C.esculentus*
Halticus lucidipennis	*C.rotundus*
Haplaxius crudus	*C.esculentus*
Heliothis virescens	*C.rotundus*
Laodelphax striatella	*C.rotundus*
Lasiglossum albescens	*C.rotundus*
Laspeyresia perfricta	*C.rotundus*
Lerema accius	*C.rotundus*
Liburniellla ornata	*C.esculentus*
Lissohoptrus brevirostris	*C.esculentus*
Locusta migratoria capito	*C.esculentus*
Macrosiphum avenae	*C.esculentus*
Macrosteles fascifrons	*C.rotundus*
Marasmima trapezalis	*C.rotundus*
Matsumuratettix hiroglyphicus	*C.rotundus*
Megelethes sp	*C.esculentus*
Megaloceroea recticornis	*C.esculentus*
Megapis dorsatta	*C.rotundus*
Melipona laeviiceps	*C.rotundus*
Micrapis florea	*C.rotundus*
Mumetopia occipitalis	*C.esculentus*
Nannobactra blepharopsis	*C.rotundus*
N. cultellana	*C.esculentus*
N. minima minima	*C.rotundus*
N. oceani	*C.rotundus*
Nephotettix nigropictus	*C.rotundus*
N. virescens	*C.rotundus*
Nomia andrenina	*C.rotundus*
N. westwoodii	*C.rotundus*
Nymphula depunctalis	*C.rotundus*
Orthoperus sp.	*C.esculentus*
Oryctes rhinoceros	*C.rotundus*
Oscinella sp.	*C.esculentus*
Oxya velox	*C.rotundus*
Pachynematus corniger	*C.esculentus*
Paraphlepsius abruptus	*C.rotundus*
Peregrinus maidis	*C.rotundus*
Phalactris politus	*C.esculentus*
Phenacoccus solani	*C.rotundus*
Pleurophorus sp.	*C.esculentus*
Pseudaletia unipuncta	*C.rotundus*
Rhizoecus cacticans	*C.rotundus*
Rhopalosiphum maidis	*C.esculentus*
R. padi	*C.esculentus*
R. rufiabdominalis	*C.rotundus and C.esculentus*
Sanctanus sanctus	*C.esculentus*
Sanctanus fasciatus	*C.rotundus*

Schizaphis cyperi	*C.esculentus*
Schizaphis siniscirpi	*C.rotundus*
Schizaphis rotundivendris	*C.esculentus*
Sitobion hillerislambersi	*C.esculentus*
Sibariops confusa	*C.esculentus*
Spathosternum prasiniferum	*C.rotundus*
Spissistilus festinus	*C.esculentus*
S. frugiperda	*C.rotundus and C.esculentus*
Sphenophorus callosus	*C.esculentus*
S. cariosus	*C.rotundus and C.esculentus*
S. destructor	*C.esculentus*
S. parvulus	*C.esculentus*
S. phoeniciensis	*C.rotundus*
S. scoparius	*C.rotundus*
S. venatus	*C.rotundus*
S. zeae	*C.esculentus*
Spodptera exempta	*C.rotundus*
S. ornithogalli	*C.rotundus*
Stenocranus rufiliearis	*C.rotundus*
Stenomicra angustata	*C.esculentus*
Stenoscinis atriceps	*C.esculentus*
Stilbus apicalis	*C.esculentus*
S. pallidus	*C.esculentus*
Syrphus sp.	*C.rotundus*
Taphrocerus schaefferi	*C.esculentus*
Telephanus velox	*C.esculentus*
Tetranychus yusti	*C.esculentus*
Thaumatimyia glabra	*C.esculentus*
Thysanpotera thrips	*C.esculentus*
Toramus sp.	*C.esculentus*
Trigonorhinus stricticus	*C.esculentus*
Truxalis grandis grandis	*C.rotundus*
Truxalis sp.	*C.rotundus*

Appendix II.

Natural enemies of *C. esculentus* and *C. rotundus:* fungi

Table. Fungal pathogens feeding on *C. esculentus* and *C. rotundus*. After (Phatak, Callaway et al. 1987)

Organism	species infected
Alternaria tenuissima	*C.rotundus and C. esculentus*
Asochyta sp.	*C.esculentus*
Balansia cyperi	*C.rotundus and C. esculentus*
C. minor	*C.rotundus*
C. peribebuyensis	*C.rotundus*
Cercospora sp.	*C.rotundus and C. esculentus*
Cintractia limitata	*C.rotundus*
Claviceps cyperi	*C.rotundus*
Curvularia tuberculata	*C.esculentus*
Drechslera mydis	*C.esculentus*
F. lateritium	*C.rotundus and C. esculentus*
Fusarium oxysporum	*C.rotundus and C. esculentus*
P. cyperi	*C.rotundus*
P. philippinensis	*C.rotundus*
Phyllachora cyperi	*C.rotundus*
Phyllosticta zingiberi	*C.rotundus*
Phytophtora cyperi+ rotundati	*C.rotundus*
Piricularia higginsii	*C.rotundus*
Puccinia canaliculata	*C.rotundus*
Puccinia conclusa	*C.rotundus*
Puccinia romagnoliana	*C.rotundus*
R. bataticola	*C.rotundus*
Rhizoctonia solani	*C.esculentus*
Sclerotinia homoecarpa	*C.esculentus*
Ustilago scitaminea	*C.rotundus*
Verticillium dahliae	*C.esculentus*

Appendix III.

Natural enemies of *C. esculentus* and *C. rotundus:* nematodes, bacteria and virus

Table. Pathogens (Nematodes, bacteria and viruses) feeding on *C. esculentus* and *C. rotundus*. After (Phatak, Callaway et al. 1987)

	Organism	species infected
Nematodes	*Criconemoides onoensis*	*C.esculentus*
	Heterodera cyperi	*C.rotundus and C. esculentus*
	Heterodera marioni	*C.esculentus*
	Heterodera mothi	*C.rotundus*
	Hoplolaimus columbus	*C.rotundus and C. esculentus*
	M. incognita	*C.rotundus*
	Meloidogyne graminicola	*C.rotundus and C. esculentus*
	Pratylenchus brachyurus	*C.rotundus and C. esculentus*
	Rotylenchus similis	*C.rotundus*
	Trichodorus spp.	*C.rotundus and C. esculentus*
Bacteria	*Azobacter*	*C.rotundus*
	Xanthomonas oryzae	*C.rotundus*
Virus	*V. lucerne dwarf virus*	*C.esculentus*