Chapter I

INTRODUCTION

Pulses are important world food crops because they provide an inexpensive source of vegetable dietary protein. In many densely populated areas of the world, the economy does not support large scale production and utilization of animal protein. In those areas, the protein in people’s diets may be augmented by supplementation with the protein rich pulse grains. In addition to being less expensive than animal protein, pulse grains provide a source of rich protein for those people who prefer vegetable to animal protein in their diet for cultural and religious reasons. Pulse grain proteins nutritionally complement the proteins in cereal grain; when consumed together, a diet nutritionally balance in protein may be enjoyed.

Green gram (*Vigna radiata* (L.) Wilczek) is one of the most important pulse crops. It is grown in almost all parts of the country. Green gram is an excellent source of high quality protein. It contains about 25 per cent protein, which is almost three times that of cereals. It supplies protein requirement of vegetarian population of the country. The protein is comparatively rich in lysine, an amino acid deficient in cereal grains. Sprouted green gram seeds provide a succulent and nutritious vegetable, rich in protein, minerals and vitamins, and available in all seasons of the year. The proximate constituents in seeds and sprouts as reported by Adams (1975) have been
converted to a moisture free basis and are given in Table 1.1. It is consumed in the form of split pulse as well as whole pulse, which is an essential supplement of cereal based diet. Among the pulses, green gram is favoured for children and the elderly due to its easy digestibility and low production of flatulence. Being a short duration crop it also provides an excellent green fodder to the animals. In addition to being an important source of human food and animal feed, green gram also plays an important role in sustaining soil fertility by improving soil physical properties and fixing atmospheric nitrogen. It is a drought resistant crop and suitable for dryland farming and predominantly used as an intercrop with other crops.

**Origin and History**

De Candolle (1986) believes that green gram has originated in India. According to Vavilov (1926) also green gram is a native of India and central Asia. It is grown in these areas since pre-historic period. Zukovskij (1962) is of opinion that *Vigna sublobata* which grows wild from India to Indonesia is the progenitor of green gram. Jain and Mehra (1978) reported that *Vigna sublobata* is not the ancestor of green gram but it appears to be so close with green gram that some taxonomists have described it as *Vigna radiata* var. *sublobata*.

**Distribution**

Green gram is grown throughout the southern Asia including India, Pakistan, Bangladesh, Sri Lanka, Thailand, Cambodia, Vietnam, Indonesia, Malaysia and China, etc. It is also grown in the parts of Africa and U.S.A. and has recently been introduced in Australia.
Table 1.1. Nutrient constituent in 100g of mature, raw seeds and sprouted seeds of green gram (moisture free basis)

<table>
<thead>
<tr>
<th>Nutrient constituent</th>
<th>Unit</th>
<th>Raw seeds</th>
<th>Sprouted seeds</th>
</tr>
</thead>
<tbody>
<tr>
<td>Water</td>
<td>%</td>
<td>0</td>
<td>0</td>
</tr>
<tr>
<td>Food energy</td>
<td>Cal</td>
<td>384</td>
<td>313</td>
</tr>
<tr>
<td>Carbohydrates</td>
<td>g</td>
<td>67.5</td>
<td>58.8</td>
</tr>
<tr>
<td>Protein</td>
<td>g</td>
<td>27.1</td>
<td>33.8</td>
</tr>
<tr>
<td>Lipids (fat)</td>
<td>g</td>
<td>1.46</td>
<td>1.77</td>
</tr>
<tr>
<td><strong>Minerals:</strong></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Calcium</td>
<td>mg</td>
<td>132</td>
<td>169</td>
</tr>
<tr>
<td>Phosphorus</td>
<td>mg</td>
<td>380</td>
<td>570</td>
</tr>
<tr>
<td>Potassium</td>
<td>mg</td>
<td>1150</td>
<td>1990</td>
</tr>
<tr>
<td>Iron</td>
<td>mg</td>
<td>8.6</td>
<td>11.6</td>
</tr>
<tr>
<td>Sodium</td>
<td>mg</td>
<td>6.7</td>
<td>45.2</td>
</tr>
<tr>
<td><strong>Vitamins:</strong></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>A</td>
<td>IU</td>
<td>89</td>
<td>177</td>
</tr>
<tr>
<td>Thiamin</td>
<td>mg</td>
<td>0.42</td>
<td>1.16</td>
</tr>
<tr>
<td>Riboflavin</td>
<td>mg</td>
<td>0.23</td>
<td>1.16</td>
</tr>
<tr>
<td>Niacin</td>
<td>mg</td>
<td>2.91</td>
<td>7.08</td>
</tr>
<tr>
<td>Ascorbic acid</td>
<td>mg</td>
<td>6</td>
<td>169</td>
</tr>
</tbody>
</table>

Adams (1975)
Area and Production

In India, green gram is grown over an area of 30.08 thousand hectares with a production of 10.23 thousand tonnes in 2000-2001 (Anonymous, 2001). The major green gram grown states are Orissa, Maharashtra, Andhra Pradesh, Rajasthan, Madhya Pradesh, Bihar, Karnataka, and Uttar Pradesh. Orissa stands first in area, production as well as productivity of green gram in India, followed by Maharashtra and Andhra Pradesh, in area and production.

In Assam, it is grown during *kharif* and summer season in an area of about 0.07 thousand hectares with a production of 0.034 thousand tones in 2000-2001 (Anonymous, 2001).

Botanical description

Green gram belongs to the family Papilionaceae (Fabaceae). It is a small herbaceous annual plant growing to a height of 30 to 120 centimeters with a slight tendency to twining in the upper branches. The central stems are more or less erect while side branches are semi-erect. The leaves are 5-10 cm long trifoliate with long petioles. Both the stems and leaves are covered with short hairs. The pods are linear, sometimes curved, round and slender with short pubescence. The seeds are small and nearly globular. The colour of seed is usually green, but yellow brown or purple brown seeds also occur. The colour of cotyledons is yellow. The crop is fully self-fertile and self-pollinated.
Soil

Green gram is grown on a variety of soils. Loam to sandy loam soils are considered ideal for green gram cultivation. Soil should be well drained since temporary water logging may damage the crop. Saline-alkali and extremely acidic soils are not suitable for green gram cultivation.

Climatic Requirements

Green gram is grown in summer and kharif season in northern India. In southern India, it is also grown in winter season. It requires hot climate and has the capacity to tolerate moisture stress. Areas with annual rainfall of 50-70 cm are considered the best for green gram cultivation. It can be grown successfully up to an elevation of 2000 m from mean sea level. Green gram can be grown within a mean temperature range of about 20 to 40°C. It is sensitive to low temperature and is killed by frost. Mean temperature of 20°C may be the minimum for productive growth, with mean temperatures in the range of 28 to 30°C being optimum.

Manures and fertilizers

Around 4–5 t of compost or farm-yard manure is required for 1 ha of land. This is to be applied at the time of land preparation. For rhizobium non-inoculated crop 32 kg of urea and 220 kg SSP is required for an area of 1 ha and in rhizobium inoculated crop 22 kg of urea and 220 kg SSP is required. In some of parts of Assam 17 kg MOP is recommended for an area of 1ha. All the fertilizers should be applied at the time of sowing.
Water management

Rainy green gram does not require irrigation unless there is a prolonged drought. Drainage of excess water from the field is essential for successful cultivation of green gram. Rabi and summer green gram may be grown only in areas where adequate irrigation facilities are available. In general, 3-5 irrigations are required for green gram in summer season.

Weed control

Green gram being a dwarf statured crop suffers to great extent if the weeds are not controlled in time. The problem of weeds is more severe in *kharif* as compared to rabi and summer seasons. Traditionally weeds are removed by mechanical means. One hand weeding after 20-25 days of sowing followed by another after about 20 days may be sufficient. Weeds can also be controlled with the use of herbicides. Pre-emergence herbicides may give good result.

Harvesting and yield

The crop should be harvested when most of pods turn black in colour. Delay in harvesting may cause shattering of pods. After sufficient drying in the sun the produce may be threshed and seeds should be cleaned. The yield of green gram in rainy season is quite low, ranging from 5-7 q/ha. The cultivation of summer green gram is an economic proposition as it gives a seed yield of 10-12q/ha which is quite remunerative at the present price level.
Pest and diseases

A large number of pathogenic fungi, bacteria, viruses and plant parasitic nematodes can infect green gram viz., web blight (Pythium sp. and Rhizoctonia solani), cercospora leaf spot (Cercospora canescens), dry root rot and stem rot (Macrophomina phaseolina), anthracnose (Colletotrichum capsici), bacterial leaf spot (Xanthomonas phaseoli), yellow mosaic virus (YMV), powdery mildew (Erysiphe polygoni), root-knot nematode (Meloidogyne spp) and the crop is also affected by several insect pests like Jassid (Empoasca kerri), white flies (Bemisia tabaci), bihar hairy caterpillar (Diacrisia oblique), beetles (Madurasia obscurella) etc.

Damage by root-knot nematode (Meloidogyne spp.)

The crop green gram is subjected to attack by many plant parasitic nematodes, out of them root-knot nematode, Meloidogyne spp. has the principal significance. Among the various species of Meloidogyne, M. incognita is considered as one of the economically important nematode pests of green gram causing serious yield losses. Mohanty and Mahapatra (1994) recorded 33.33 to 37.77 per cent loss in yield of green gram due to M. incognita in Orissa. This nematode has been found to be one of most destructive in green gram cultivation as reported by several workers (Singh, 1972; Chahal and Chahal, 1987). Considerable works have been done on various aspects of management of this nematode on different crops (Bora and Phukan, 1983; Bhattacharya and Goswami, 1988; Prasad and Mittal, 2004; Rajendran and Saritha, 2005; Ravishankar and Singh, 2005), but very limited work has so far been
carried out pertaining to this nematode on green gram (Sakhuja and Singh, 1980; Ray and Dalei, 1998; Gogoi and Neog, 2003; Borah et al., 2007).

The pathogen, root-knot nematode (*Meloidogyne* spp.)

The root-knot nematodes, a common name collectively given to the species of *Meloidogyne* causing knots (galls) on the roots of wide variety of plants, are undoubtedly the most well known and extensively experimented nematodes. These are prevalent in menacing proportions in most parts of the world, especially in the sub temperate, subtropical and tropical regions and are considered to be the number one nematode problem of agricultural crops in the most developing nations. Collectively, the species of root-knot nematodes are considered among the top five of the major plant pathogens and one or more of the species attack nearly every crop responsible for the world’s supply of food, fibre, timber, resins, ornamentals or other cash crops (Sasser, 1979). Among the species present in India, *Meloidogyne incognita* and *Meloidogyne javanica* have the widest host ranges covering over 232 and 144 genera of plants in India respectively (Krishnappa, 1985). Many more hosts have been recorded thereafter. The more preferred hosts are dicot vegetables, pulses, fibre, fruit and cash crops (Plate 1 a-i). Some monocots like sugarcane, pearl millet, banana etc. are also heavily attacked. These species often creates serious problems in nurseries of fruit trees/vines, vegetables and ornamentals. Certain crops, though normally non-hosts, may be attacked in heavily infested soil, for instance.

The root-knot nematodes are, generally, more abundant in sandy and sandy loam soils with 50 per cent or more sand. The juveniles are more abundant in
the upper 20 cm layer of the field soil but vertical distribution may be influenced by temperature and moisture changes. Populations are relatively higher in deeper layers under too high or low temperature or high moisture stress.

The root-knot nematodes are basically parasites of roots or underground stem or pods etc. The above ground symptoms hence are those of slow debility of root in its function of nutrient and water uptake and translocation. The plants may be dwarfed yellowish with smaller foliage. The below ground symptoms on the roots are small galls which in case of multiple infection on the near by tissues, may coalesce to form large galls. The general appearance of the crop in a field is of patchy growth due to uneven distribution of the nematode. The poorly growing patches grow in size each year and spread more in the direction of ploughing and irrigation water. The symptoms advance with the age of the crop and more spectacular under drought or low fertility levels. Usually, the injury is not detectable from the aboveground symptoms until the crop has already suffered almost 25 per cent damage. Both, quality and quantity of produce are affected.

**Life cycle of root-knot nematode**

The eggs are laid in a gelatinous matrix which generally protrudes out of the host tissue but at times may be embedded in it. The contents of the egg undergo a series of determinate cleavages giving rise to the blastula and triploblastic gastrula stages and finally the first stage juvenile. The first moult occurs within the egg to give rise to the second stage juvenile that is ready to hatch out. The hatching can occur freely in water or moist soil without any requirement of root exudates or
Plate 1 (a-i). Economically important crops infected by root-knot nematode
hatching factors. However, under adverse conditions of moisture, osmotic pressure, temperature etc., the hatching is inhibited or postponed to ensure longer survival.

The pre-parasitic second stage juvenile (Plate 2) moves freely in soil in search of a suitable host tissue. The juvenile has sufficient glycogen and glycolipid reserves for survival for several months in case the host is not available. These are also capable of surviving under adverse conditions.

The juveniles feed on the epidermal cell of the underground plant parts and penetrate the newly formed tissue such as above the meristematic zone. They usually orient parallel to stele and with head pointed opposite to the tip. The nematode may be found attached to the more mature roots since the root apex grows further.

The juvenile usually starts feeding on the pericycle cells. The cell contents are liquefied and semi digested extra corporeally with the help of hydrolytic enzymes secreted by oesophageal gland. The nematode enzymes also induce excessive conversion of tryptophan into indole acetic acid and certain other changes. This results in enlargement (hypertrophy) and coalescing of pericycle cells into a group of multinucleate giant cells around the nematode head. The cortical parenchymatous cells around the giant cell undergo excessive multiplication (hyperplasia) giving rise to tiny swellings on the roots, or primary galls, several of which may merge into big multiple galls.

The post infectional second stage juvenile continues to feed for a period of several weeks, gradually swelling and losing the capacity to move. The period of feeding (2-3 weeks optimally) varies with the host reaction, temperature, moisture and
other environmental factors. A series of three moults occur in quick succession. The cuticles of second, third and fourth stages are retained till the last moult. Hence, these stages have the projecting tail cuticle (spike) of the second stage. In most species a majority of juveniles develop into pyriform sedentary females (Plate 3) while the rest produce monorchic males. Under environmental extremities proportionately more males are produced and some of the juveniles destined to be females actually produce diochoric males.

The reproduction may be parthenogenetic or bisexual. The female lays about 400-500 eggs into a gelatinous matrix secreted by the rectal glands. The total number and size of eggs are affected by host status and the level of environmental stress. The total duration of life cycle under optimum conditions is 3-4 weeks in most species. However, the duration is affected by a number of ecological factors especially host status, temperature, moisture, pH, osmotic pressure etc. Tyler (1938) found that the duration from juvenile to egg laying female was the least (17 days) at optimum temperature of 27.5-30 °C but longer up to 57 or more days at low and high temperature.

**Root-knot nematode in Assam**

The first record on the occurrence of the plant parasitic nematodes in Assam was made in the Annual Report of The Tocklai Experimental Station in 1949 as eel worms associated with tea plant. Das (1958) recorded the occurrence of root-knot nematode, *M. incognita, M. hapla* and species of *Pratylenchus* on roots of tea seedlings. After that several workers reported the occurrence of root-knot nematode on
Plate 2. Second stage juveniles of *Meloidogyne incognita*

Plate 3. Adult females of *Meloidogyne incognita*
different host plants from Assam and adjoining North East India (Phukan et al. 1981; Phukan and Sarmah, 1983; Bordoloi and Phukan, 1987; Choudhury and Phukan, 1992).

The root-knot nematode, *M. incognita* is of common occurrence in almost all pulse and vegetable growing areas of Assam and has been posing a serious threat to the cultivation of many crops including green gram. In Assam yield losses due to *M. incognita* ranged from 17.14-56.64 per cent was recorded on various economically important crops like green gram, black gram, okra, soybean, french bean, pea, lentil, jute, carrot etc. (Das, 1992; Das, 1994; Das and Phukan 1987; Deka, 1996; Deka and Rahman 1997; Devi and Das 1995; Hazarika, 1996; Kalita, 1988; Saikia and Phukan 1986).

**Control measures**

During the last decade, considerable works have been done on various methods of control measures to minimize yield losses caused by plant parasitic nematodes. Among them application of nematicides has been found to be very effective in controlling nematode pests of different crops, but its frequent use may induce residual effect, phytotoxicity and health hazards to human and livestock. The major problem of nematicide application is the ground water contamination particularly by halogenated hydrocarbons and carbamate group of nematicidal chemicals. These chemical pesticides can be toxic to non–target organisms and they damage the functioning systems where they are used (Rajgopal et al., 1984). The retention time of these chemicals in soil is still higher and they are considered to be
influence the beneficial microorganisms in soil (Phukan and George, 1991). Judicious use of nematicide has not taken place under Indian condition due to lack of awareness by many farmers. With the growing awareness of limitation of nematicides in agriculture, approaches in Integrated Nematode Management have come out to be more encouraging and ecofriendly. Biological control of nematode is one of the most important components of Integrated Nematode Management programme and thus emphasis has been focused on biological control of plant parasitic nematodes by exploitation of biological agents in view of greater awareness of pollution free environment.

Biological control is becoming a necessary component for safe and effective plant diseases management. With increasing knowledge and concern about effects of chemical pesticides on the environment, many chemicals may no longer be available. Its have the potential to fill the gap created by the disappearance of the broad spectrum pesticides.

Among the various fungal antagonists identified so far for biological suppression of plant parasitic nematodes causing economic damage on agricultural crops, the potential role of vesicular arbuscular mycorrhizal (VAM) fungi has been the subject of intensive study in recent years in an effort to develop ecologically sound nematode management strategies (Jalali and Jalali, 1991).

VAM fungi are associated with a greater variety of plant species and are more widely spread geographically than other types of mycorrhizal fungi. These endophytes have a wide host range and thus are common to most cultivated crops as
well as to natural plant communities (Gerdemann, 1975). Despite this universality, VAM had not been investigated extensively until the last decade. This oversight came primarily because they have little effect on root morphology and the fungal endophytes cannot be grown in pure culture. However, improved research techniques and proof of the role of VAM in plant nutrition have stimulated great interest in this symbiotic association. The VAM fungi produce characteristic structures known as vesicles and arbuscules inside the root and form obligate beneficial symbiosis in the roots of most terrestrial plants (Trappe, 1987). Arbuscules occur within the cortical cells of the plant roots and vesicles occur within or between them. These two structures are used to distinguish VAM from other fungi present in the rhizosphere. These two structures together with intraradical coenocytic hyphae constitute the vegetative phage of VAM fungus in the root and are important in the acquisition of carbon and nutrients for vegetative growth (Harley and Smith, 1983; Bowen, 1987).

VAM fungi invade the primary cortex, vascular tissue, the secondary cortex and the thick fleshy roots. The VAM fungi infect a plant root either from a germinating spore, infected root piece containing intraradical spores or intra or extra radical hyphae present in soil. Prior entering into the host cells, the germinating spores produce an appresorium on the epidermal cells, subsequently, the fungus invade the cortical cells through the epidermal layer and produce hyphae, arbuscules and vesicles. In general, the production of arbuscules precedes production of vesicles during the growth cycle of the fungus. Arbuscules are formed by dichotomous branching and are abundant in the cortical cells (Kiden and Brown, 1975). Arbuscules are short lived and their life vary from one to three weeks, after which it is digested (Bowen, 1987). The
arbuscules are the most significant structure of VAM fungi and are the preferential site for fungus/plant metabolic exchange (Cox and Tinker, 1976). Vesicles are globose body; develop by an intercalary or terminal swelling of VAM hyphae in the root cortex. They contain many lipid droplets and serve mostly as storage organ inside the roots (Bonfante Fasolo, 1984). This group of fungi contributes substantially to the establishment, productivity and longevity of natural man made ecosystem. They don’t exhibit host specificity and even a single species can infect and establish symbiotic relationship with a diverse group of plant species in nature.

VAM fungi are classified based on morphology and germination characteristic of asexual spores under the division of Eumycota, class Zygomycetes, order Endogonales, and belongs to the family Endogonaceae (Trappe, 1982). In the soil these fungi produce such structures as azygosporles, chlamydospores, sporocarps, vesicles and mycelia. VAM fungi colonize plant roots and project an extensive network of hyphae into the surrounding soil, thus increasing the absorptive area of the root system for nutrient and water uptake. The relationship aids primarily in the absorption and translocation of phosphorus, an ion of low solubility and immobile in the soil. Other elements such as sulfur, copper and zinc are sometimes involved. Hence, by increasing the supply of phosphorus and nutrient to the plant the VAM fungi stimulate growth and in return receive organic nutrients from the host. VAM fungi are also important for their potential application as biofertilizer in agriculture which influence growth and development of plants by increasing uptake of less mobile nutrients like phosphorus, zinc and copper by plant especially from low fertility soil (Bolan, 1991). The introduction of promising VAM fungal isolates into arable fields
offer one possible method of developing sustainable agriculture for increasing the uptake of residual immobile phosphorus. Thus, the use of VAM fungi enhances utilization of fertilizers immediately applied to a crop and those fertilizers that exist as residual from previous application to the field soil (Cox and Rabson, 1980). Mycorrhizal plant grow better in infertile soil largely because fungus hyphae can obtain poorly mobile nutrients from beyond the zone of nutrient depletion surrounding roots in soil, and indeed for this reason many plants with short or rudimentary hairs depend more on mycorrhiza. The fungal hyphae propagate the association and interconnect plants. Hyphae can also influence soil structure by helping to produce humic acid, weathering soil minerals and stabilizing large soil aggregates. Mycorrhizal roots are considered to be more efficiently respond to spatial and temporal variations in soil nutrient supply (Brundret, 1991). Therefore, it is not only useful as biofertilizer but these mycorrhizal fungi have also been shown to enhance water transport in plants, decrease transplant injury, help plants withstand high temperature, increase drought resistance of plant, promote establishment of plants in wasteland and reduce the vulnerability to diseases caused by soil borne pathogens (Safir et al., 1972; Shonbeck, 1979; Nelson and Safir, 1982; Graham, 1986).

VAM fungi and plant parasitic nematodes are commonly found inhabiting together in the roots or rhizosphere of the same plant, each having a characteristic but opposite effect on plant growth. The obligatory symbiotic VAM fungi may stimulate plant growth, whereas the obligate plant parasitic nematodes usually suppress plant growth (Hussey and Roncadori, 1982). It has been observed that the plants heavily colonized with mycorrhizal fungi are able to grow well in spite of
the presence of damaging levels of plant parasitic nematodes (Hasan and Jain, 1991; Jain and Hasan, 1994). VAM have an antagonistic effect on plant parasitic nematodes, and such an effect have either a physiological or a physical basis. An interaction could have a physical basis if the endophytes and endoparasitic nematodes in the root compete for the same site, rendering it unfavourable for nematode activities. A negative interaction occurs when plant growth or yield of dually infected plants is less than that of unchallenged mycorrhizal plants or if root colonization or sporulation by the mycorrhizal fungus is suppressed.

Many mechanisms operate simultaneously during interaction of VAM and plant parasitic nematode on a particular host. Following factors play major role in reducing nematode population and in enhancing plant growth.

i) VAM fungi may increase root growth, expand the absorptive capacity of the root system for nutrients and water and enhance cellular process in roots (Hayman, 1982).

ii) VAM fungi improve plant nutrition and by doing so may aid the host in compensating for damage caused by parasitic nematodes, thereby increasing plant tolerance to the pathogens.

iii) Mycorrhizal root colonization has been shown to affect root exudation (Gerdemann, 1968; Harley and Smith, 1983; Hayman, 1982). These changes could alter chemo tactic attraction of nematodes to roots or directly retard nematode development within root tissues.
iv) VAM fungi have reduced nematode infection and development on several hosts in spite of larger root systems on mycorrhizal plants (Cooper and Grandison, 1986; Grandison and Cooper, 1986; Smith et al., 1986a). It may be due to unfavorable conditions for nematodes development on root system due to VAM colonization. Change in hormones, amino acids and cell permeability in roots have been attributed to mycorrhizal symbiosis (Hayman, 1982). Thus, VAM fungi may inhibit nematode activities by altering the root physiology in ways unrelated to P nutrition and enhanced root growth.

v) VAM fungi may cause change in the post infection nematode host interaction by altering nematode reproduction and development.

vi) Direct competition for space and available photosynthates produced in the plant may account for reduced nematode infection on mycorrhizal root system since endoparasitic nematodes occupy similar root tissues as VAM fungi (Saleh and Sikora, 1984; MacGuidwin et al., 1985; Smith et al., 1986a).

vii) VAM colonization of roots mechanically preventing nematode penetration and establishment (Umesh et al., 1988).

viii) Increased phenol and decreased auxin levels in the mycorrhizal infected root impede larval growth and giant cell formation (Dehne et al., 1978).

ix) Greater accumulation of coumestrol in mycorrhizal infected roots. Six weeks after inoculation of VAM results activated metabolism involving de novo synthesis of enzymes by which plants infected by VAM are more resistant to subsequent attack by nematodes (Morandi and Bailey, 1984).
In addition to above given factors, increased tolerance or resistance to nematodes by VAM fungi may be due to production of nematistatic compounds and parasitism of eggs. More likely, VAM fungi alter the host either physically or physiologically, and thus indirectly affect the host-nematode relationship (Smith, 1987).

Several findings indicate that VAM fungi able to increase plant growth and yield, reduce nematode penetration, development and increase nutrient uptake specially phosphorus in several host plant (Roncadori and Hussey, 1977; Sitaramaiah and Sikora, 1982; Sivaprasad et al., 1990a; Jothi and Sundarababu, 1997; Sreenivasan et al., 2003). It was also found that VAM fungus, *Glomus fasciculatum* was effective in reducing penetration and delayed the development of different stages of *M. incognita*. It also increases plant growth parameters and yield of various pulse crops including green gram (Sankaranaryanan and Sundarababu, 1994; Ray and Dalei, 1998; Mahanta and Phukan, 2000).

Apart from VAM fungi some other bacterial and fungal biocontrol agents such as *Pasteuria penetrans*, *Trichoderma harzianum*, *T. viride* and *Paecilomyces lilacinus* are commonly used for the management of several plant parasitic nematodes and has been established as promising biocontrol agents for the management of root-knot nematode (Stirling, 1984; Sharma and Trivedi, 1989; Somasekhar and Gill, 1991; Walia et al., 1991; Rao et al., 1997a; Cannayane and Sivakumar, 2001; Devi and Sharma 2002; Gogoi and Neog, 2005).
In the last few years, considerable research work has been conducted on exploitation of VAM fungi with other management approaches against root-knot nematode *M. incognita* (Jain and Hasan, 1995; Krishna Rao *et al.*, 1995; Sankaranarayanan and Sundarababu, 1997) but very limited works in this direction have so far been conducted in Assam. Therefore, to develop a sensible, rational and effective integrated management strategy for root-knot nematode, the present investigation was undertaken with the following objectives to study the -

1. Influence of different spore levels and time of inoculation of *G. fasciculatum* for the management of *M. incognita* on green gram.

2. Compatibility of *G. fasciculatum* with neem based pesticides as seed treatment for the management of *M. incognita* on green gram.

3. Compatibility of *G. fasciculatum* with organic amendments and carbofuran for the management of *M. incognita* on green gram

4. Interactive influence of *G. fasciculatum* with biocontrol agents of *M. incognita* infecting green gram.