SICKLEPOD

Sicklepod (*Senna obtusifolia*) in Queensland

PEST STATUS REVIEW SERIES - LAND PROTECTION

By
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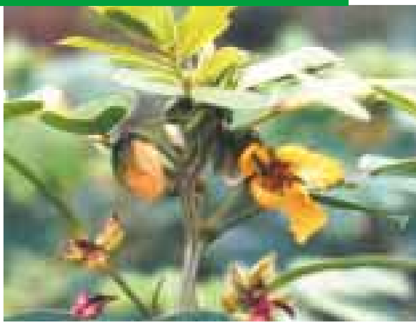

Queensland Government
Natural Resources and Mines

Acknowledgements

The authors wish to thank the many people who provided information for this assessment. In particular, landholders, land managers, Local government and National Parks staff gave generously of their time when discussing the impacts and costs of sicklepod. A special thanks goes to Graham Hardwick, Peter van Haaren, Ron Kerwyk, Rick Beattie, Marie Vitelli, Graham Schultze, Peter Austin and Andrwe Mitchell for more technically based advice and information.

Cover and contents design:
Grant Flockhart and Sonia Jordan

Photographic credits:
Natural Resources and Mines staff

ISBN 0 7242 7258 5
Published by the Department of Natural Resources and Mines, Qld.

Land Protection
Department of Natural Resources and Mines
Locked Bag 40, Coorparoo Delivery Centre, Q, 4151

Contents

1.0 Summary

Sicklepod is found along the tropical east coast of Queensland from Cape York to just south of Mackay and often grows in association with a closely similar species, foetid cassia. The two species are not commonly distinguished in weedy situations and field based information on sicklepod probably also refers to foetid cassia. It is likely that sicklepod currently infests approximately 600,00 ha in north and far north Queensland. Ecoclimatic modelling suggests that sicklepod could spread along the entire east coast of Queensland and foetid cassia could grow across the base of Cape York and into the country around the Gulf of Carpentaria.

Sicklepod is a weed of pastures and of sugar cane. It has the potential to become a major weed of several other crops. In pasture, it is an aggressive invader and can completely dominate grass species, eradicating pasture growth and excluding stock. Carrying capacities can be reduced by as much as 85%. It is conceivable that properties could become completely unproductive. Although it is generally unpalatable to stock, if eaten, sicklepod is toxic to cattle. If left to grow in a sugar cane crop, sicklepod can have a significant effect on yield. Processing contaminated cane can cause machinery breakdowns at sugar mills because of the woody nature of sicklepod. It is also causing concern as an environmental weed in some native ecosystems in Far North Queensland. The economic impact of sicklepod to Queensland has not been fully established, but current control costs are estimated to be $00000 annually.

Sicklepod is dispersed by water (stream flow and floods), in mud on machinery and stock, in mulch and by stock which ingest the pods. In cane areas it is also transported by cane bins on tramways and harvesters. Seed production is profuse (8000 seeds per plant) and as the seed is long lived, an abundant seed bank can become established in the soil. Scarification of the seed is required for germination to occur, and sometimes fires can cause mass emergences of seedlings because of this. The plant grows on a variety of soil types and is usually an annual, although it can perenniate.

Sicklepod is difficult to control by chemical means. Spraying should be carried out at the seedling stage for the most effective control. Slashing delays seeding, but does not kill the plant (unless blunt blades are used to shatter the stem) and may cause it to perenniate. Ploughing often leads to an increase in plant numbers as the seeds are scarified during ploughing. Biological control is not available, although field work to identify possible agents has been carried out.

The current areas of sicklepod infestation should be mapped as an aid to formulating control strategies and the rate of spread must be contained. Research on control methods must be extended. Because of its impact, sicklepod may be a good candidate for a major coordinated control programme, although some characteristics of the plant mitigate against this.

2.0 Taxonomic Status

Previously known as *Cassia obtusifolia* L. it was placed in the genus Senna by Irwin and Barneby (1982). A second species, *Cassia tora* L. or foetid cassia, sporadically grows in association with sicklepod in Australia and is commonly mistaken for it. Irwin and Barneby (1982) did not consider *C. tora* in their revision, since it does not occur in the New World, and this species remained, inappropriately, in the genus *Cassia*. However Randell (1988) accepted the use of three genera to replace *Cassia sensu lato* and in consequence placed *C. tora* in the genus *Senna* (Appendix A). The correct names are thus *Senna obtusifolia* (L.) Irwin and Barneby (sicklepod) and *Senna tora* (L.) Roxb. (foetid cassia).

Sicklepod belongs to the subfamily Caesalpinioideae of the family Leguminosae and, together with foetid senna, is placed in the Section *Chamaefistula* along with several other introduced *Senna* species (appendix A). *Chamaefistula* also contains two native *Senna* species, but other Australian *Senna* species are placed in the section *Psilorhegma* and *Senna.*

2.1 Description

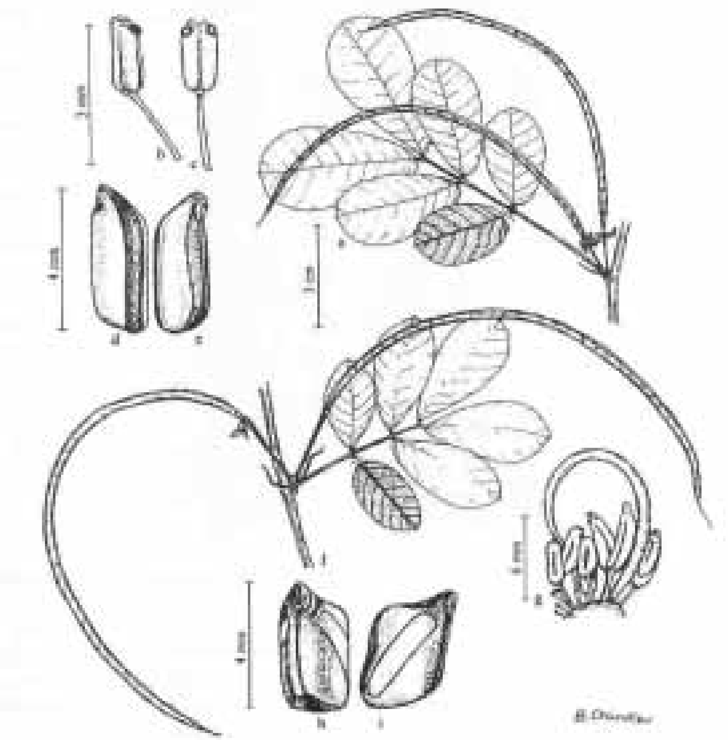

Fig. 1. Sicklepod and foetid cassia - general morphology: a, *S. tora* pods and compound leaf; b & c, *S. tora* anthers showing truncated tips; d & e, *S. tora* seeds showing the broad longitudinal areole; f, *S. obtusifolia* pods and compound leaf; g, *S. obtusifolia* - part of flower showing the three large beaked anthers and the recurved style with its tip positioned over the anthers; h & i, *S. obtusifolia* seeds showing the narrow transverse areole (modified after Randell 1988).

Detailed descriptions of sicklepod have been given by Irwin and Barneby (1982) and Randell (1988) who also provides one for foetid cassia. Both species are non-nodulating legumes (Parsons and Cuthbertson 1992, Randell 1988) and do not have nitrogen fixing bacteria associated with roots (Waterhouse and Norris 1987). More general descriptions of sicklepod are given by Flint *et al*. (1984), Anning *et al*. (1989), Anon. (1989) and Hall and Vandiver (1996). The following is a general description based upon these authors which applies to both species.

Annual or short-lived perennial herbs or somewhat woody subshrubs 0.5-2 m tall; stem erect and nearly hairless, compound leaves, including petiole 4-6 cm long, alternate, with 2-3 pairs of obovate leaflets increasing in size from the base of the rachis, the largest of which are 2-5 cm long by 1-3 cm wide, veins conspicuous on the leaf undersurface, leaf green to dark green with a paler undersurface; 1-2 erect glands between the lowest leaflet pairs; inflorescence 1-2 flowered, flowers 0.8-1.0 cm across, usually in pairs from the upper leaf axils; peduncle 3-4 mm long; bracts caducous[1]; sepals unequal, elliptic; petals unequal, yellow, bluntly rounded at apex; stamens 10, comprising 3 small infertile ones, 4 fertile normal ones and 3 larger fertile ones on elongated filaments; ovary covered with fine hairs; fruiting peduncle 2-4 mm long; pod curved and initially green but turning dark brown with maturity (Figs. 1 & 2).

Fig. 2. A flowering stem of sicklepod (*Senna obtusifolia*) showing the flower head, seed pods and shape and structure of the leaves (Source: Hall and Vandiver 1996).

The seedling of sicklepod (Fig. 3) (Hall and Vandiver 1996) has rounded cotyledons 15-20 mm broad, which are green above and light green below and which have 3-5 distinct veins in the upper surface joining the mid vein. The stem appears to be almost smooth but in fact is covered with short downward-pointed hairs.

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[1] caducous: falling off easily.

Fig. 3. The seedling of sicklepod (*Senna obtusifolia*) showing the two broad, rounded cotyledons (Source: Hall and Vandiver 1996).

2.2 Distinguishing Characters

Sicklepod and foetid cassia have often been confused but Brenan (1958) noted that the two species are clearly distinct and suggested that they could be reliably distinguished on the basis of the shape of the anthers and the seed areole (*tora*: areole on side of seed 1.5-2.0 mm broad; *obtusifolia*: areole on side of seed 0.3-0.5 mm broad). The number of glands (extra-foliar nectaries - Retzinger 1984) between the lower leaflet pairs has often been used to distinguish the two species but this is not a constant character for separation of the two species according to Singh (1978) who confirms the usefulness of Brenan's characters and suggests that the seed testa[2] is also a useful distinguishing feature. In *S. obtusifolia* the testa is slightly muricated[3] and not distinctly veined, but in *S. tora* it is not muricated but is distinctly veined. The separation of sicklepod and foetid cassia as distinct species is supported by Upadhyaya & Singh (1986) who noted that there was no natural hybridization when both species were grown in adjacent plots. They also noted that the two species differ in respect of several phytochemical characters.

Randell (1988) separates *S. obtusifolia* from *S. tora* on the following basis:

> S. *obtusifolia*: petioles 1.5-2 cm long; fruiting pedicels 2-3 cm long; anthers with short beaks; seed areole narrow, not longitudinal.
>
> S. *tora*: petioles 2-4.5 cm long, fruiting pedicels to 1.5 cm long; anthers truncate, beakless; seed areole broad, longitudinal.

Sicklepod and foetid cassia are distinguished from other Australian members of the genus *Senna* with cylindrical pods by having 1-3 pairs of leaflets and by the seeds having areoles (Randell 1988). In the field, *S. tora* can be distinguished from *S. obtusifolia* by the foetid smell which arises from its crushed foliage (Waterhouse and Norris 1987).

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[2] testa: the protective outer covering of the seed.
[3] muricate: covered by short, hard-pointed protuberances.

Since *S. tora* appears to have lost several phytochemical constituents possessed by *S. obtusifolia* (Upadhyaya and Singh 1986) it may have evolved from *S. obtusifolia* (Cock and Evans 1984). There are two forms of *S. obtusifolia* (Irwin and Barneby 1982): that found in the USA has a uniglandular extra-floral nectary on the upper surface of the rachis between the two lower leaflets (and 2n=28). This originates from the Caribbean. Another form from British Guiana, Surinam and Venezuela has two extra-floral nectaries (and 2n=26) and a relatively narrower pod. Randell (1995) suggests that *S. tora* is derived from the broad-podded variant of *S. obtusifolia* which is found in the Antilles and USA. Tandon and Bhatt (1971, cited in Upadhyaya and Singh 1986) have shown the two species are caryologically distinct even though both have *n*=13 and suggest that as there are forms of *obtusifolia* known which have *n*=12, 13, 14 and some Indian forms of *tora* have *n*=13, 14, so *tora* could have arisen via aneuploid loss from *obtusifolia*. It is not clear which form of *S. obtusifolia* is present in Australia although Randell (1988) gives pod diameter as 0.3 cm which is in the range of (2-)2.5-3.5 mm diameter given by Irwin and Barneby (1982) for the narrow podded variety.

3.0 History of Introduction and Spread

Senna tora was first recorded in Australia in 1871 but the earliest collection was made at Port Darwin in 1888 (Symon 1966). It established in Queensland in 1917-18 as an escapee from green manure trial plots (Symon 1966). *S. tora* is only sparingly naturalised in Queensland and is not a declared plant. *S. obtusifolia* appears to have been introduced to Australia in the Second World War around Darwin (Parsons and Cuthbertson 1992). It was first recorded as a weed from the Northern Territory in 1961 and from Queensland in 1963 (Randell 1988). It was declared as a noxious plant in Queensland under the *Rural Lands Protection Act* on 17 October, 1981.

4.0 Current and Predicted Distribution

4.1 Distribution - overseas

Sicklepod probably originated in the Neotropics (Irwin and Barneby 1982, Flint *et al*. 1984, Parsons and Cuthbertson 1992) but now has a pan tropical distribution. It is found in Africa (tropical west, central and east Africa, Namibia and South Africa) India, Sri Lanka, Pakistan, Malaysia, the Philippines, Indonesia, Papua New Guinea, USA (including Hawaii), and below 1650 m in Mexico and Central America, the Caribbean and from Colombia to Brazil to Paraguay to Argentina (Irwin and Barneby 1982, Randell 1995). It is found throughout most of the eastern and southern states and it is a signficant economic impact where it grows in crops, particularly leguminous ones, such as soybeans. The range of sicklepod in the USA is similar to that of 150 years ago. However, it is increasing in infestation within this range but is not invading other areas of the USA (Teem *et al*. 1980). However, there is evidence that the evolution of tolerance to cooler temperatures may be occurring at the northern limits of its current range (Patterson 1993). If this is correct it has clear implications for the eventual increase in distribution of sicklepod in Australia, particularly if the cold tolerant biotype is already present in the country.

S. *tora* is restricted to the old world, principally from the Indian subcontinent eastwards and it is likely that the species evolved in the Asia-Pacific region (Randell 1988). Only a single specimen is known from Africa, which could have been introduced (Brenan 1958). *S*. *tora* is found in Arabia (particularly the monsoonal coastal strip of Oman) Pakistan, India and east through southern China Thailand, Cambodia, Laos, Vietnam, Malaysia, Singapore, Indonesia, the Philippines, New Guinea and Australia to Polynesia (Singh 1978, Randell 1995, Boufford, 1996 *pers. comm.*, Gardner 1996 *pers. comm.,* Lock 1996 *pers. comm.*, Veldkamp 1996 *pers. comm.*). Within Polynesia, it is found in the Solomons, American Samoa, Western Samoa, Fiji, Guam, Tonga, Vanuatu and the Futuna Islands (Waterhouse and Norris 1987). It is also recorded from Jamaica where it may be a recently introduced exotic (Cock and Evans 1984).

4.2 Distribution - Australia

The known distribution of sicklepod in Queensland is shown in Fig. 4. It is found from Bamaga in the far north through the Atherton Tablelands to the central coast south of Mackay, in grazing and sugar cane regions which get more than 1650 mm rain p.a. (James and Fossett 1982, Anon. 1989).

Fig. 4. The distribution of *Senna obtusifolia* in Queensland (dark grey: heavy infestation; hatched: scattered infestation; Queensland Herbarium records: * naturalised plant; + cultivated plant).

In north Queensland extensive and extremely dense infestations occur, but in Central Queensland infestations tend to be light and scattered although heavy infestations occur south of Proserpine in the Andromache and O'Connell River catchments and around the upper part of Atherton Creek near Mareeba. Its distribution in sugar cane areas is centred on Innisfail, the Johnstone River, East Palmerston (south-east of Milaa Milaa) and Ingham (Symon 1966) and it is spreading around Tully and Ingham (Anon. 1989). In the Mackay region in cane areas, it is generally found on headlands. It spreads most rapidly on hills: possibly because of the better drainage, greater seed flow and heavy grazing in the wet season (Anning *et al*. 1989). In the Northern Territory sicklepod is an important weed of land just rising from the flood plain of coastal rivers. It is found through Arnhem Land, south to Katherine and west to the Daly River basin (G. Schultze, *pers. comm.* 1996).

Foetid cassia is found sporadically in the Mackay, Cairns and Innisfail areas (Fig. 5) (James and Fossett 1982/83, Ablin 1996 *pers. comm*.) but its detailed distribution and the level of infestation is not known. Elsewhere in Australia it appears to be restricted to the Northern Territory in the Darwin region (Parsons and Cuthbertson 1992) but as in Queensland, where sicklepod infestations are found, *S. tora* is not generally discriminated from *S. obtusifolia*. A single infestation in Western Australia was recently been discovered at a quarantine wash-down facility at Kalumburu (A. Mitchell *pers. comm.* 1996).

Fig. 5. The distribution of *Senna tora* in Queensland (Queensland Herbarium records: * naturalised plant; + cultivated plant).

Based on information suppled by the Department of Natural Resources and Mines' Land Protection Officers, sicklepod currently (1996) infests approximately 600,000 ha in north and far north Queensland. The range expansion of sicklepod, indicated by the effective radius of the infestation which is measured as √Area (Shigesada and Kawasaki 1997) is shown in Fig. 6. It appears to be following the course of the Type 3 range expansion of Shigesada and Kawasaki (1997) which is characteristic of organisms with a long distance dispersal mechanism which allows the establishment of nascent populations, and their independent increase. A situation which seems to adequately describe the sicklepod invasion which is still in rapid expansion phase in Queensland. One way to reduce the rate of range expansion suggested in Fig. 6 would be to halt long distance dispersal using the mechanisms discussed in Section 7.1.

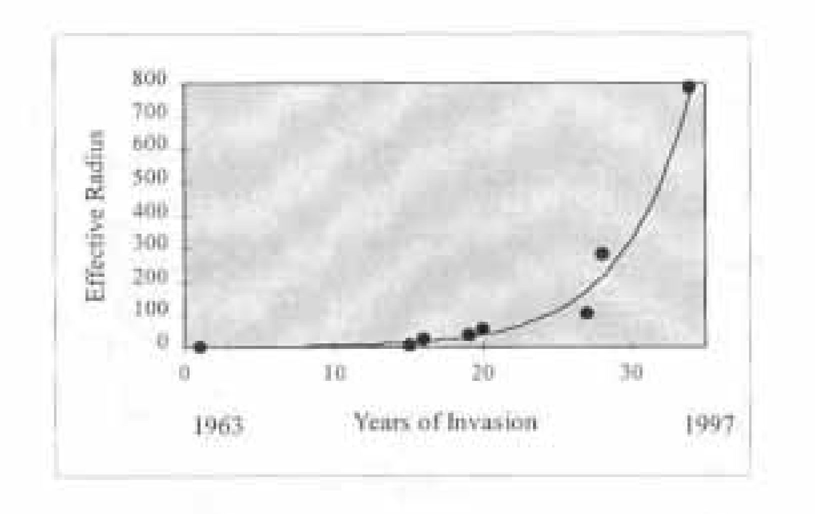

Fig. 6. The rate of spread of sicklepod in Queensland predicted to the year 2000. Data from Anning *et al.* (1989) and NR&M staff.

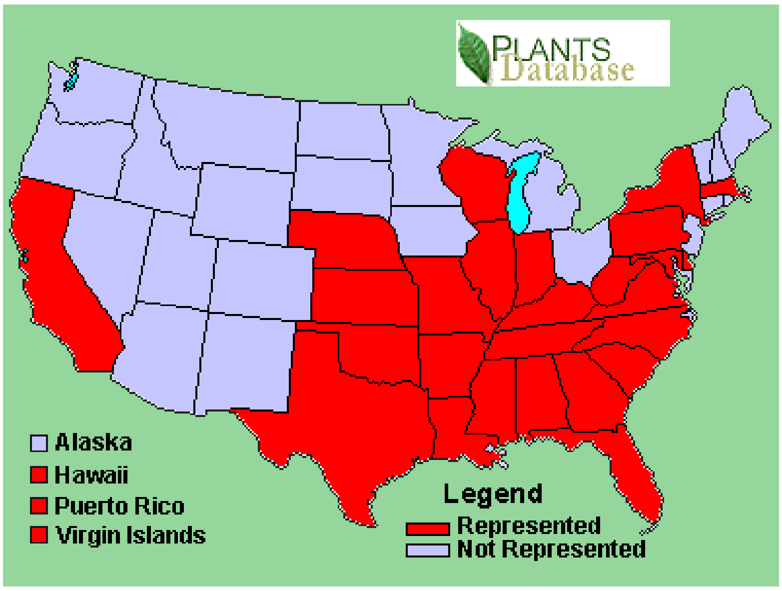

Fig. 7. The distribution of sicklepod (*Senna obtusifolia*) in the USA.

CLIMEX (Skarratt *et al*. 1995) was used to model the potential distributions of sicklepod and foetid cassia in Australia based on the ecoclimatic characteristics of the regions in which the species is thought to be native. The distribution used as the basis for the sicklepod model was that of the species in the USA (Fig. 7). The distribution of foetid cassia in south-east Asia (Fig. 8) was used in modelling the distribution of this species.

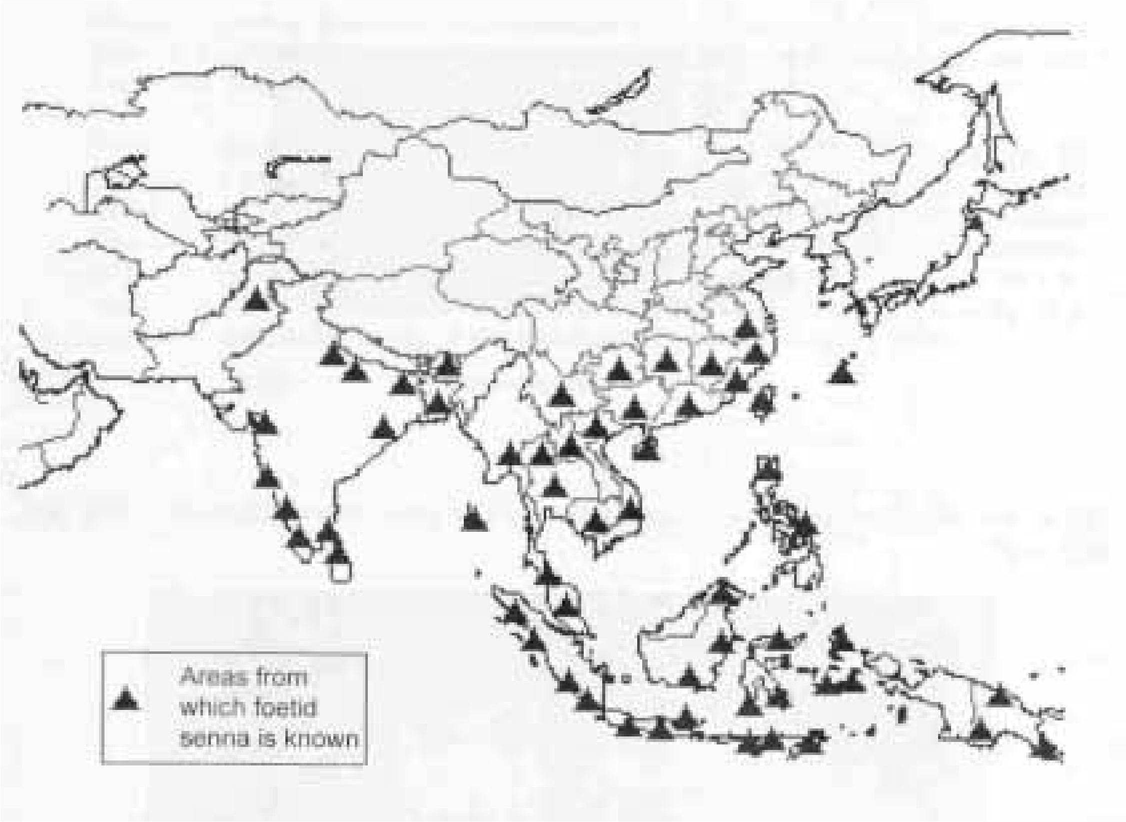

Fig. 8. The distribution of foetid cassia (*Senna tora*) in Asia

The ecoclimatic model for sicklepod predicted the North American distribution of sicklepod well (cf. Figs. 7 & 9) but predicted the presence of this species in Illinois and Indiana, where it has not apparently been recorded. However, Farnsworth and Bazzaz (1995) used sicklepod seed collected from wild populations in Illinois when assessing the effects of elevated carbon dioxide levels on plant reproduction and growth and so it seems possible that sicklepod could also occur in Indiana. The model also predicted the known pantropical distribution of the weed well (Fig. 10).

Although sicklepod is generally thought of as a tropical and subtropical weed in Australia (Parsons and Cuthbertson 1992) the model predicts that it should be able to grow right along the eastern coastal strip of Australia (Fig. 11). Because the model is based on the wide climatic tolerances exhibited by sicklepod in the USA, the distribution of the plant in Australia is unlikely to be restricted by temperature extremes in Australia and the model predicts that the distribution in Australia is most likely to be limited by soil moisture availability and dry stress. Although sicklepod has a potentially widespread distribution in Australia its optimal habitat is predicted to be coastal Queensland and New South Wales.

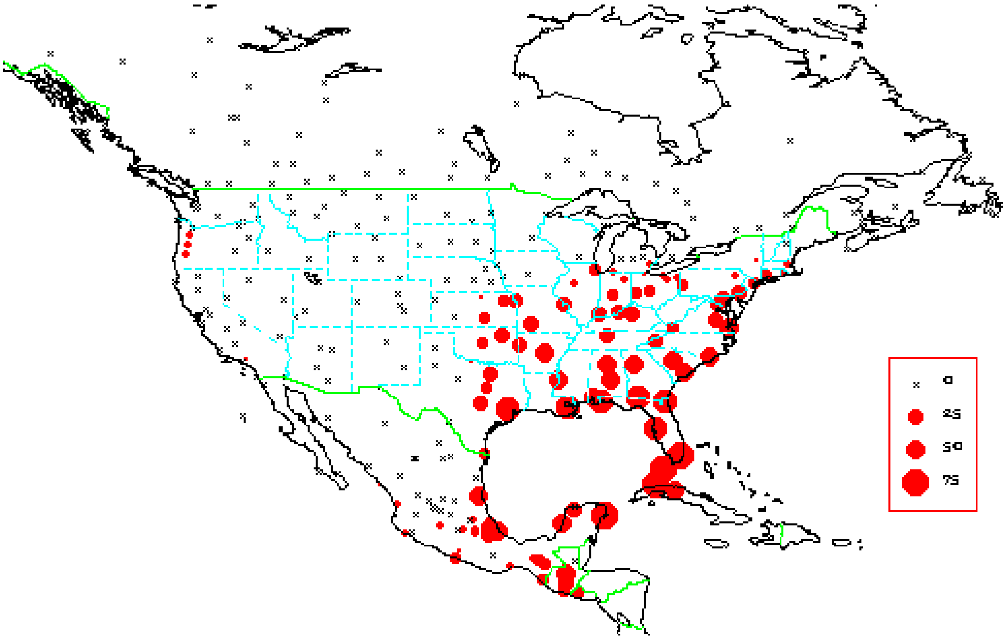

Fig. 9. The predicted distribution of sicklepod (*Senna obtusifolia*) in the USA (EI = Environmental Index, a measure of the potential of a location to support a population of a species).

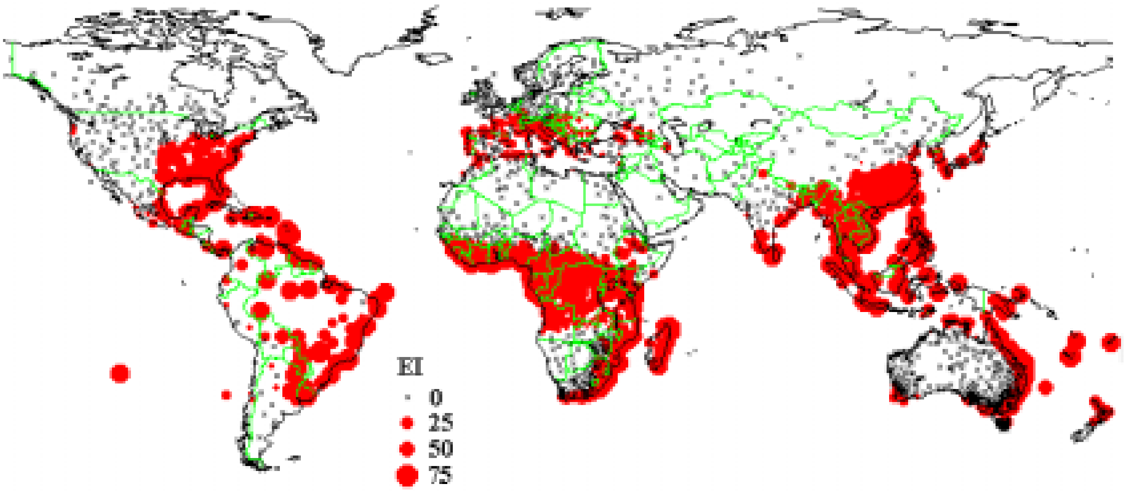

Fig. 10. The predicted world distribution of sicklepod (*Senna obtusifolia*) (EI = Environmental Index, a measure of the potential of a location to support a population of a species).

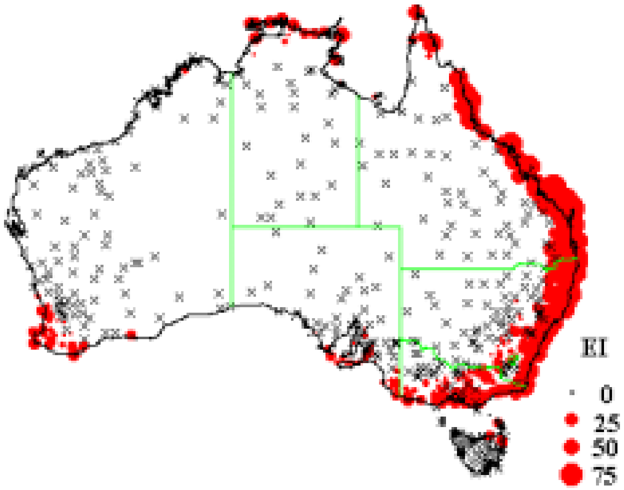

Fig. 11. The predicted distribution of sicklepod in Australia (*Senna obtusifolia*) (EI = Environmental Index, a measure of the potential of a location to support a population of a species).

The ecoclimatic model for foetid cassia predicted the Asian distribution of this species well (Fig. 12) and also predicted the Pacific distribution of the species well (Fig. 13). The predicted distribution within Australia (Fig. 14) was more restricted than for sicklepod, reflecting perhaps, a loss of tolerance for a wide range of ecoclimatic factors during the evolution of *S. tora* from *S. obtusifolia* as evidenced by the narrower tolerance of *S. tora* for variation in seed storage temperatures (Singh 1968). Notwithstanding, the models indicate that of the two species, foetid cassia should be able to grow in the Kimberley region of Western Australia and interestingly, subsequent to the development of these models, the first record for either of the two species in this region was established and the plant was reliably identified as *S. tora* (A. Mitchell *pers. comm.* 1996).

5.0 Estimates of Current and Potential Impact

5.1 Impact on Primary Industry

5.1.1 Detrimental

Both species are weeds of pastures and crops. In Australia, sicklepod is a vigorous and unpalatable weed that produces a dense cover (Anon. 1989, Anning *et al*. 1989). The relative contribution of foetid cassia to sicklepod stands in Australia is not known, but it is a major pest of pastures in the Pacific, probably as a response to over grazing (Cock and Evans 1984). It is the worst pasture weed in Vanuatu and is a major weed in Fiji and Tonga (Waterhouse and Norris 1987). In Australia, all 'sicklepod' infestations are attributed to *S. obtusifolia*. In Queensland, sicklepod is primarily a weed of pastures where it reduces available grazing areas, competes with pasture for light, nutrients and water and can rapidly exclude all other species. In the Mackay area some sicklepod paddocks now carry one third of the cattle they used to (Anning *et al*. 1989) and *Queensland Country Life* (23 April 1992) reported that carrying capacity on one property near Cooktown was increased from 1 animal / 8 ha to 2.5/ha after up to $700/ha was spent on sicklepod control. Sicklepod is a major problem in the Northern Territory, particularly along the margins of floodplains where it quickly invades any suitable habitat.

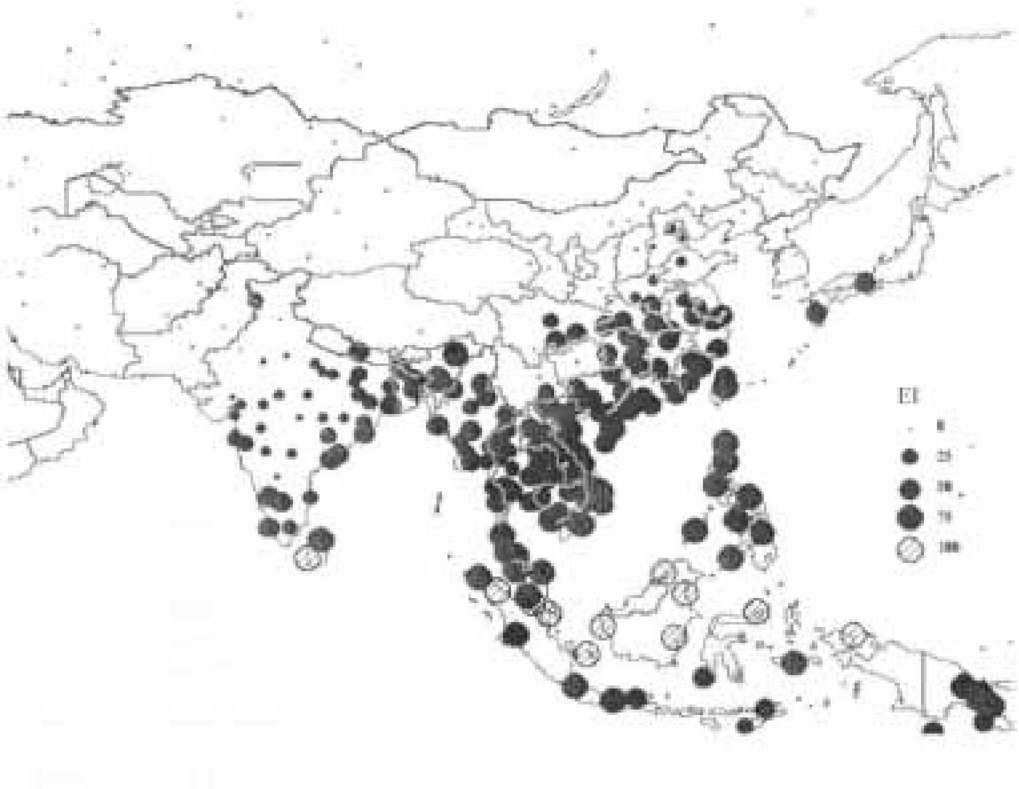

Fig. 12. The predicted Asian distribution of foetid cassia (*Senna tora*) (EI = Environmental Index, a measure of the potential of a location to support a population of a species).

Sicklepod is generally unpalatable to cattle, but if eaten is quite toxic (Nicholson *et al*. 1985/86, Anon. 1996b). Animals are poisoned through eating the plant in the field, in green chop, hay, or if the seed is contaminating grain. Confined yearling cattle are most likely to eat the weed, especially if the weed is common and there is little other forage. Ingestion rates of 5.5-11.5 kg per adult dairy cow will cause poisoning within 4 to 10 days of ingestion (Nicholson *et al*. 1985/86). The toxic principles are not known, although the related *S. occidentalis* (coffee senna) which produces similar symptoms on ingestion, contains anthraquinone glycosides which cause diarrhoea (Dowling and McKenzie 1993). The toxins appear to be concentrated in the seed although they are also present in the leaves and stem. They appear to act on the skeletal muscles, kidney and liver (Anon. 1996b).

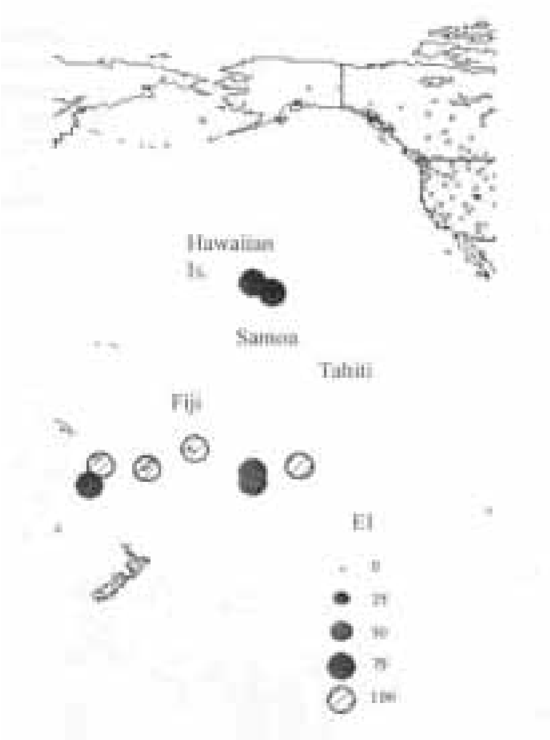

Fig. 13. The predicted Pacific distribution of foetid cassia (*Senna tora*) (EI = Environmental Index, a measure of the potential of a location to support a population of a species).

The first symptom of sicklepod poisoning to appear is usually diarrhoea. Animals then go off their feed, appear lethargic and then tremors begin in the hind legs indicating muscle degeneration. Segmental degeneration of the muscle fibres is caused although the sarcolemmal membrane and the nuclei remain distinct (Nicholson *et al*. 1985). As this degeneration progresses, the urine becomes dark and coffee coloured because muscle damage liberates myoglobin to the urine (Nicholson *et al*. 1985) the animal becomes recumbent and cannot rise. Death occurs within 12 h of the animal going down.

Treatment is usually ineffective once the animal is recumbent. Selenium and vitamin E injections have been used as treatments with variable results (Anon. 1996b). No records of stock death from sicklepod poisoning have been recorded in Australia (Parsons and Cuthbertson 1992). Dogs may have been poisoned after eating meat from a steer poisoned by the closely related *S. occidentalis* (coffee senna). Post-mortem changes caused by sicklepod poisoning are likely to be similar to those described for coffee senna by Dowling and McKenzie (1993). The limb muscles are pale and there are scattered pale areas in the heart muscle; haemorrhage in the muscle of the heart and congestion of the lungs are commonly found.

Stock do not eat *S. tora* (Lock 1996 *pers. comm.*) although they are reputed to eat it in green silage and as the dry seed pods (Cock and Evans 1984). Nothing is known of its toxicity.

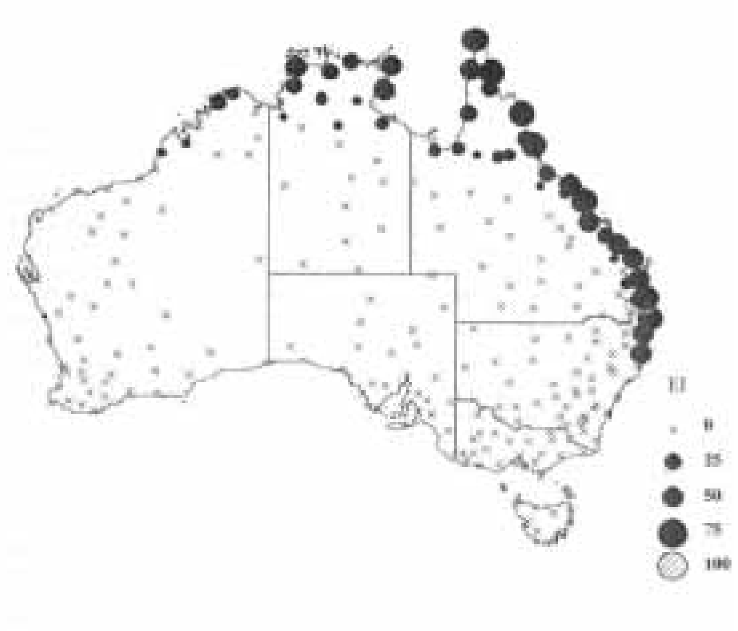

Fig. 14. The predicted distribution of foetid cassia (*Senna* tora) in Australia (EI = Environmental Index, a measure of the potential of a location to support a population of a species).

In Australia, sicklepod is also a weed of sugar cane (Anon. 1989) and is probably the most serious weed of cane in the Herbert River District (Kerkwyck 1996, pers. comm.) infesting about 25,000 ha of the 55,000 ha of cane in the region (Beaty 1996, pers. comm.). Early reports suggested it would only grow up to 12 rows into the crop (James & Fossett 1982/83) but more recent reports indicate that it usually grows throughout the crop. Sicklepod competes vigorously with the cane for light, moisture and nutrients and increases the difficulty of cane harvesting.

Although sicklepod and foetid cassia together produce significant weed infestations in pastures and sugar cane stands in Queensland, it is unlikely that they have reached their crop weed potential It has been suggested that sicklepod is allelopathic (Waterhouse and Norris 1987, Anon. 1989) and there have been reports of the growth of signal and pangola grasses being suppressed for 2-3 years after sicklepod control has been affected (J. Arnold, L. Warren *pers. comm.* 1997).

Foetid cassia is a pest of coconut and pigeon pea (Cock and Evans 1984) in the Pacific and sicklepod is a pest of cotton, soybeans and peanut in the southern USA and of groundnuts in West Africa. Although sicklepod is considered less competitive than many broadleaf weeds (Anon. 1996c) in 1984 in 6 southern US states sicklepod rated in the top ten most troublesome weeds in sorghum, soybeans, and corn in five states, and peanuts in four states and was reported as the fifth most troublesome weed and sixth most common weed in the southern US (Flint *et al*. 1984). A more recent survey (Webster and Cobble 1997) has shown that sicklepod has increased in importance in the southern USA and now ranks as first, fourth, fifth and seventh most important weeds in soybeans, peanuts, corn and cotton respectively. Teem *et al.* (1974, cited in Retzinger 1984) suggest increasing sicklepod populations are probably due to herbicide resistance but Webster and Cole (1997) indicate that sicklepod's long-season discontinuous germination pattern allows it to exploit areas where competition from other weeds has been removed by herbicide application.

At a density of 16-32 sicklepod plants per 15 m of row, seed cotton yield was reduced by 50%. The percentage of trash in cotton increased with sicklepod density and mechanical harvesting was affected at high densities (Buchanan *et al*. 1978). At densities of 0.5 sicklepod plants per metre of row, soyabean yields in Florida can be reduced by 20% (Currey *et al.* 1981) and 10 plants m^{-2} can reduce groundnut yield by 6-22 kg ha^{-1} (Hauser *et al.* 1982). Sicklepod is a prohibited noxious weed in Virginia (Abaye 1992) and contamination of seed lots (of soybean) by sicklepod seeds is considered a serious problem as contaminated seed cannot be sold. Contamination of seed also occurs in North Carolina and Tennessee: 5% of seed lot samples tested were contaminated with sicklepod seed (Whitt 1992). Whilst sicklepod is not yet an established crop weed in Queensland, the situation in the southern states of the USA indicates that sicklepod could establish as a crop weed in Queensland and have a significant impact on yields and associated control costs.

5.1.2 Beneficial

Neither sicklepod nor foetid cassia have a beneficial impact in Queensland, although in Nigeria and the Cameroons sicklepod has many medicinal uses (Hutchinson and Dalziel 1937 cited in (Cock and Evans 1984)). The root especially has purgative and antihelminthic properties and the leaves are used to treat skin diseases. In the Phillipines foetid cassia is used as purgative and vermifuge and in Indo-China it is used as a medicine for dysentery and opthalmia (Quisumbing 1951 cited in Cock and Evans 1984).

5.2 Economic Costs

There is a general lack of information on the economic and environmental impacts of sicklepod. This is a characteristic shared by many weeds. The following examples provide a cross section of the economic impacts that sicklepod has on three different systems being; sugar cane production, beef production and National Parks and Indigenous areas. A more detailed assessment would be required to provide an estimate of the full impact of sicklepod on the Queensland economy.

5.2.1 Sugar cane

The management cycle of sicklepod control in sugar cane generally conforms to the following pattern. Sicklepod is introduced onto a property by one of several means. These include contract cane harvesters, contract dozers for earthworks and/or flooding. A few plants are noticed the following year but do not appear to be a problem (sicklepod is sometimes initially mis-identified as other less problematic weeds). However, due to prolific seed production, sicklepod spreads rapidly and can reduce cane yields within 2-3 years of introduction. Landholders then initiate more effective control measures to combat sicklepod, such as timely and sufficient chemical control. Diligent control is required for many years and seed production has to be prevented to deplete the seed bank. Gradually sicklepod lessens as a problem.

Chemical applications are required to control sicklepod in sugar cane and these can be coordinated to provide control of other crop weeds. Control costs vary depending on the stage at which sicklepod is sprayed. Anning *et a/.* (1989) indicate that sicklepod is easier and less expensive to kill when plants are small. Small plants can be controlled for about \$35 ha^{-1} but larger plants may cost up to \$60 ha^{-1}. Tordon 75-0 ® is temporarily registered for control of sicklepod in sugar cane. It can take more than one spraying per year to control sicklepod.

There are currently about 55,000 ha of sugar cane in the Herbert River area of which an estimated 25,000 ha are infested with sicklepod. Weed control is generally effected by a chemical mix such as Gramoxone®, 2,4-D and Diuron® for about \$40 ha^{-1}. However, if sickle pod is tall enough, it requires the application of Tordon 75-0® at a cost of about \$60 ha^{-1}. The difference in spray costs of \$20 ha^{-1} is attributed to sicklepod. Assuming 80% of the infested area is ratoon cane and requires spraying with Tordon 75-0®, then 20,000 ha x \$20 ha^{-1} = \$400,000 expenditure directly attributable to sicklepod. While this is a very rough estimate, it provides an indication of the potential magnitude of the control costs of sicklepod in sugar cane. Chemical application costs have not been included here because the cane would have to be sprayed for weeds regardless of sicklepod.

If it is left to grow in the crop, sicklepod can have a significant effect on cane yields, even though sugar cane is a strong competitor. Weeds in general can reduce cane production by 13-30% if left uncontrolled (BSES 1989), and although this was not specific to sicklepod, some production loss from sicklepod can be expected. Further costs arising from sicklepod in the crop are increased cane harvesting costs and contamination of harvested cane. The latter, when excessive, has been known to cause breakdowns at sugar mills due to the woody nature of the weed.

Sicklepod is spread along rail lines as infested cane is transported to the mill. This requires extra spraying to control the weed in these areas in addition to normal weed control practices. Sicklepod also needs to be controlled on the non-cropped areas of farms such as headlands, riparian areas and around farm buildings.

Table 1 provides a summary of the estimated chemical costs per year of sickle pod in those cane areas between Ingham and Innisfail with known sickle pod infestations. Other mill areas either had no, or negligible amounts of sicklepod. Contract spraying, by ground or aerial application, costs up to \$30 ha^{-1} over and above chemical costs, and needs to be considered when determining the costs of sicklepod. These costs have been included in the estimates in Table 1, where appropriate (but not in the Herbert, see above).

Table 1. Estimated chemical costs per year attributed to sickle pod in selected cane areas.

Mill or cane area	Chemical costs in 1996/7
Herbert	\$400,000
Tully	\$120,000
Mourilyan	\$40,000
South Johnstone	\$30,000
Mulgrave	\$20,000
Total	**\$610,000**

Sicklepod is becoming recognised in almost all the areas mentioned in Table 1 as an increasing problem and this suggests that the amount of chemical control could double or treble in coming years.

These preliminary estimates indicate that the current cost of sicklepod to the sugar industry in Queensland would be over \$600,000 y^{-1} and that this mainly comprises the chemical costs of control and application costs. The Herbert area with estimated costs of \$400,000 dominates the total. It is estimated that over 90 percent of growers with sicklepod present on their properties in the Herbert area are actively controlling it (R. Beattie, *pers. comm.* 1997).

5.2.2 Beef

Sicklepod is a weed of degraded pastures and especially old sugar cane fields where it can totally suppress pasture production. Sicklepod also flourishes on the more fertile flats; the most productive areas of a property. Estimates of the impact of dense infestations of sicklepod range from reductions in carrying capacity from 66% (Anning *et al.* 1989 and Queensland Country Life, 23 Apri11992) to almost 100%.

While sicklepod is known to be toxic to stock, it has not been an economic issue to date because stock tend not to eat it, preferring other vegetation to sicklepod. No cases of poisoning have been reported in Australia.

Case Study – A beef property in north Queensland

A case study approach was undertaken of a property in northern Queensland that has a dense sicklepod infestation. The current costs of sicklepod comprise control costs and reduced beef production. Control slashing and spraying costs over \$11,000 y^{-1} and consist of labour, chemicals, spraying equipment and machinery usage. There are about 300 ha of dense sicklepod infestations and about 600 ha of scattered infestations. The dense infestations are slashed whilst the sicklepod has immature pods. This prevents regrowth but does not necessarily encourage pasture growth. The scattered infestations are treated by foliar applications of herbicide.

There is no pasture production in the dense infestations (e.g. Fig. 15) and pasture production in the scattered infestations is reduced to about 5 percent. Over the property this converts to loss of about \$20,000 in gross beef production each year.

Therefore, total losses in term of control costs and forgone beef production is in the order of about \$31,000 y^{-1}.

Sicklepod can be controlled by herbicide but some areas, such as heavily timbered paddocks, islands in water courses and other riparian areas may be difficult or impossible to control. In these situations sicklepod tends to dominate pastures and may become a monoculture. Sometimes landholders may not stock these paddocks, even if there is available feed, because of the risk of spreading sicklepod to other parts of the property.

A major problem currently inhibiting the control of sicklepod on beef properties is the poor state of the beef industry and the subsequent poor financial performance of beef properties in general. Landholders often have insufficient financial resources to control the weed.

A survey of beef properties would be required to accurately estimate the total impact of the weed on the beef industry, although the several properties that were surveyed for this study yielded costs between \$2,000 and \$31 ,000 y^{-1}.

5.2.3 National Parks and Forestry

Sicklepod impacts on national parks and forestry areas, especially those reclaimed from old grazing properties. Although the control costs in these areas can be valued, the effects of sicklepod on the ecological values of an area cannot.

National parks, such as Lumholtz National Park near Ingham, are encountering problems with sicklepod. Expenditure on sicklepod had been as high as $15,000 to $20,000 y^{-1} and is currently about $5,000, but park staff are closely monitoring its spread and will renew efforts once a recently acquired property bordering the park has been destocked (K. Smith, *pers. comm.* 1997).

Case study – A national Park in north Queensland

An example of sicklepod causing problems in a National Park is Iron Range National Park which is about 400km of Cooktown on the Cape York Peninsular. Sicklepod has spread rapidly in the last 10 years along least 42km of the roadway in the Park and is threatening the remaining significant stands of undisturbed native grasslands in the area. It is believed that the main forms of spread through vehicles along the roadways, feral pigs disturbing the soil and via flooding along the Claudie River (M. Blackman, *pers comm*. 1997)

The annual control effort for sicklepod growing along the roadways in the park consists of foliar spraying with Grazon DS ® which requires 6 weeks labour each year. This typically costs about $6,000. Attempts to control sicklepod along the waterways involves the propagation and planting of rainforest tree species around the perimeter of infestations in an effort to prevent the spread of sicklepod and to eventually shade out the weed. The main cost of this is a half a person year in labour annually to undertake the propagation and planting. Salary, on-costs and other expenses come to about $13,000 y^{-1}.

Approximately 14,000 tourists and local visitors visit the park each year. If sicklepod infestations increase throughout the park, the number of visitations may decrease. Sicklepod is also threatening the biodiversity values and ecological integrity of the park. Likewise, sicklepod is having an increasing impact on the cultural heritage and other values of aboriginal land adjacent to the park where the weed is also present and spreading along the Lockhart River. It I difficult to identify, and value, the impacts of weed invasions on ecological values.

Dryander National Park near Airlie Beach north of Proserpine also has sicklepod present. The weed is currently threatening ecological values in the park. Dryander was formerly a forestry and grazing property so the sicklepod is concentrated around watering points such as dams, and along forestry 'snigging tracks'. Current expenditure is around $3,000 y-1 on spot spraying (including labour, chemicals and equipment usage) which mainly involves control around the infested dam. The park also undertakes strategic burning to aid control because it is not possible to spray the 'snigging tracks' due to the difficult terrain (B. Nolan, *pers. comm.* 1997).

Sicklepod is a problem in many forestry areas. For example, Department of Primary Industries (DPI) -Forestry at Ingham have been spraying sicklepod in plantations for about 7 years for a total cost of about $43,000 (in 1997 dollars). Their control program is currently costing about $6,000 y^{-1}. Similarly, DPI at Cardwell has just commenced a control program of about $10,000 y^{-1} to prevent the further spread of sicklepod in Caribbean Pine plantations in the area. The Department of Natural Resources and Mines (NR&M) - Forestry , at Ingham spends about $2,000 y^{-1} on sicklepod control in native forests.

5.2.4 Shire Councils

Many shires have indicated that sicklepod has become a major weed on public land in the last 4-10 years, and is an increasing problem. Shire councils in infested areas spent an estimated $133,000 on sickle pod control in 1996/97. A breakdown of estimated expenditure by shire is provided in Table 2. The estimated percentage of expenditure spent on sicklepod by each shire council gives an indication of the extent of the problem in that shire.

Expenditure has been similar to that in shown Table 2 for the past few years, but as sicklepod continues to spread, it may place greater pressure on council budgets. Mareeba Shire plans to spend up to $8,000 in 1997/98 on sicklepod control and Eacham Shire will probably spend at least $2,000 on monitoring and inspection. A greater amount of time will be spent in future years "encouraging" landholders to control sicklepod on private land (through education, extension and enforcement). Some shires, which have not been spending much on sicklepod, have suggested that it could become their major weed in the near future.

Table 2. Estimated Shire Council expenditure on sicklepod control 1996/7

Shire council	Estimated expenditure	Estimated percent of total weed budget
Cook	$70,000	100%
Douglas	$13,000	33%
Cairns	$1,750	4%
Herberton	$3,200	10%
Johnstone	$1,450	3%
Cardwell	$8,000	15%
Hinchinbrook	$2,000	12%
Whitsunday	$15,500	40%
Mackay	$15,000	50%
Mirani	$500	4%
Sarina	$2,500	40%
Total	**$132,900**	

5.2.5 Other

NR&M (formerly Department of Lands) undertook control research in the late 1980's and early 1990's. This mainly focussed on chemical control options and an initial exploration for bioloqical control aqents in Central and North America.

NR&M will continue to make a significant investment in biological control research on sicklepod in coming years. About $90,000 will be spent in financial year 1997/98 and $170,000 in both 1998/99 and 1999/2000 with the potential for further funding in subsequent years. About 80% of the funding is subject to annual review. These estimates include labour, overhead and operating costs.

Funding will cover overseas collecting of potential agents, introduction into quarantine and host plant testing and further overseas exploration as necessary. Any insects approved for release will be mass reared and released. Basic ecological studies of the reproductive processes of the weed and an extension and communication plan will be intrinsic to the project.

Some landholders mentioned that they have been forced to change agricultural practices because of sicklepod. For example, some have had to change from growing more lucrative small crops (horticulture) such as pumpkins and rock melons to growing sugar cane because sicklepod could not be controlled in the broad leaved horticultural crops. The forgone potential profits could not be valued but are significant.

Sicklepod has also necessitated the purchase of capital equipment to be able to undertake control. Landholders have bought spray equipment such as utility packs to spot spray sicklepod in pastures and raised tractors to spray sicklepod in taller sugar cane.

Some control work has been undertaken near the Wujul Wujul and Hopevale Aboriginal communities in the past. However, sickle pod is a growing problem on some Aboriginal managed lands and has the potential to impact substantially on traditional activities such as hunting and access to bush tucker.

5.2.6 Overall Cost Assessment

A preliminary economic assessment of sickle pod has shown that it is having a significant impact in all infested areas. Sicklepod is currently costing cane growers in Queensland at least \$610,000 y^{-1} in chemical control costs and Shire councils are spending about \$133,000 annually. It has been estimated that the impacts on infested beef properties range from about \$2,000 to \$31 ,000 y^{-1} in control costs and lost beef production.

National parks are also affected with about \$3,000 to \$19,000 being spent annually at selected national parks and sickle pod is also spreading on state forestry land and Aboriginal managed areas. NR&M and DPI at Ingham and Cardwell are currently spending about \$18,000 to control sicklepod in forestry each year. NR&M is also making a significant investment in biocontrol research of at least \$430,000 over the next 3 years, with the potential for continued research, in an effort to find effective control agents.

Based on the above estimates, it would appear that the costs of sicklepod currently is almost \$1 million y^{-1} (depending on extent of the actual impact on the beef industry). This cost is dominated by chemical control costs and should be considered as a conservative estimate. Sicklepod has the potential to get much worse in years to come, thereby significantly increasing the amount of control which will be required in future years.

Many other costs such as changed management and ecological costs could not be valued but are thought to be significant. A more complete economic assessment would be possible if the extent and density of sicklepod infestations were mapped and accurately estimated.

5.3 Environmental Costs

In areas favourable for its growth, sicklepod is capable of excluding all native vegetation and of forming dense monospecific stands, but in Queensland at least, it predominantly inhabits disturbed areas, grazing and crop lands. As such, sickle pod may be able to invade open native plant communities, and as mentioned above, it is threatening small remnant areas of native grassland in the Wet Tropics. In Dryander National Park, the open forest community is heavily impacted, and sicklepod *also* grows along old snigging tracks in vine forest on ridges (8. Nolan, pers. comm. 1997). Hardwood State Forests which have been disturbed by logging may also be susceptible to invasion. Feral cattle, feral pigs and the occurrence of natural events such as cyclone, which open up the forest canopy, are likely to enhance the possibility of invasion into pristine areas.

Whilst there is no compelling evidence to suggest that sicklepod is an aggressive environmental weed in Queensland, the information available suggests that this aspect of its impact may just not be evident at this stage of the invasion process. The environmental costs of sicklepod are therefore minimal at present but may well increase in the future.

6.0 Biology and Ecology of Spread and Control

Very little information is available on the biology and ecology of foetid cassia and the following account is based largely on sicklepod. Since both foetid cassia and sicklepod form mixed stands in Queensland and Singh (1968) observes that both species usually grow in association with each other, the information presented is probably applicable to both species.

6.1 Habitat

Both species are weeds of roadsides, waste places, open woodlands, pasture and cultivated fields (Parsons and Cuthbertson 1992, Anon. 1996c) and are adapted to a wide range of soils (Anning *et al*. 1989). Sicklepod can grow at soil pH's of 4.7 to 6.3 (Creel *et al*. 1968) but in sandy loam shows reduced growth and stunting at pH 4.7 and 5.2 (Buchanan *et al*. 1975). It has an optimum in the range of 5.5-6.0 (Creel *et al*. 1968). The stunting was perhaps due to Mn and Al toxicity at low soil pH, with the metals coming into solution in the soil (Buchanan *et al*. 1975). Even so, sicklepod seems to have medium to high pH tolerance compared to sixteen other weed species (Buchanan *et al*. 1975). Soil phosphorus and potassium affect growth (Buchanan and Hoveland 1973, Hoveland *et al.* 1976). Sicklepod is tolerant to low soil phosphorus levels but responds well to phosphorus fertilisers. At low soil potassium concentrations, growth is significantly reduced and plants become stunted.

Studies in the USA show sicklepod has a high optimum temperature for growth (Creel *et al.* 1968, Teem *et al.* 1980, Flint *et al*. 1984, Patterson 1993 and Hall and Vandiver 1996). Seed germinates in the range 18-36 ^{o}C with maximum germination rates being variously reported as between 24-33 ^{o}C and 24-36 ^{o}C. Maximum hypocotyl elongation occurs at 30 ^{o}C. Seedling growth occurs in the temperature range of 18-39 ^{o}C with a narrow optimal range around 33^{o} C. Day and night temperatures significantly affect the net assimilation rate and growth performance. As day and night temperatures increase from 23/17 to 29/23 ^{o}C plant height, leaf area, total dry weight and axillary branch leaf production increase. Plant height is at a maximum at 34/26 ^{o}C day/night. The number of main stem nodes, total leaf number and leaf area all have a maximum at 29/26 ^{o}C to 34/26 ^{o}C day/night temperatures. Warm nights in particular enhance sicklepod growth. The threshold for leaf production was around 13 ± 1 ^{o}C.

6.2 Morphology

Both species are an erect, robust shrub growing to a variable height, although *S. tora* tends to be more diffuse in habit (Singh 1968). In India at least, *S. tora* rarely grows to more than two feet (60 cm; Roxburgh (1832) cited in Singh 1968) and Singh (1968) records a maximum height of 70 cm, whereas *S. obtusifolia* can grow to 2.5 m (James and Fossett 1982) although more usually it grows to around 1.5-1.8 m (Abaye 1992, Patterson 1993). In both species plant height depends on ecotype (Singh 1968, Irwin and Barneby 1982, Retzinger 1984) and in *S. obtusifolia* at least, external factors such as temperature, summer rainfall and photoperiod: plant height is greater at higher temperatures and when summer rainfall is high (Retzinger 1984, Patterson 1993). Primary root length is sharply reduced by simulated drought and if seedlings are cut 1 cm below the cotyledons, adventitious roots do not develop and vegetative regeneration fails (Hall & Vandiver 1996). At longer photoperiods (>13h) shoot and fruit weight are greater than at shorter photoperiods (Patterson 1992). Irwin & Barneby (1982) state that when starved or crowded *S. obtusifolia* flowers as a small single stemmed plant less than 10 cm but in Queensland where the plant grows in extremely crowded stands it grows to at least 1.5 m at flowering (Fig. 14). As well as being taller and more robust than *S. tora*, shoot and root dry weight, pod length and root penetration into the soil are greater in *S. obtusifolia* (Singh 1968).

Fig. 15. A dense stand of flowering sicklepod in north Queensland.

There are two morphological types of *S. obtusifolia* found in the Americas (Irwin and Barneby 1982). The first has narrow, needle-like pods outwardly and downwardly curved (Central America and northern South America). The second has a broader, less curved pod and the seeds are compressed and obliquely tilted (not near vertical) across the pod's axis (USA and West Indies). Elsewhere the distributions of plants with these pods types often overlap.

The leaves of *S. obtusifolia* are photosensitive and the leaflets fold upward by flexible petioles at night or on cloudy days (Hall and Vandiver 1996).

6.3 Phenology

Both species are annual weeds (Waterhouse and Norris 1987, Abaye 1992) but in the wet tropics, *S. tora* will frequently perreniate. Slashing during the vegetative phase can cause sicklepod (James and Fossett 1982) and foetid cassia (P. van Haaren *pers. comm.* 1996) to become a short lived perennial. The two species may flower at different times (Vatsavaya S. Raju and N. Rama Rao 1986 cited in Randell 1995). In Queensland, sicklepod will germinate and grow at any time of the year if the weather is warm and sufficient rain has fallen. However, the main germination period is in mid-summer after the summer rains have started. During growth, many leaves are shed so that by the start of flowering in late summer, much of the main stem is bare. Flowering continues through the autumn and pods appear and then ripen until midwinter. The plant dies back in late winter and spring.

6.4 Floral Biology

The duration of the vegetative phase of sicklepod depends on ecotype and climate and varies from 43-84 days before the first flower appears (Retzinger 1984). Flowers of the Cassiinae usually do not secrete nectar as the pollen is shed through short slits or pores in the anthers and the pollen is released through the vibration of the flowers by bees during 'buzz pollination' (Gottsberger and Silberbauer-Gottsberger 1988). Many species of *Senna* are well adapted to buzz pollination but in *S. obtusifolia* the gynoecium is recurved over the anther pores (Fig. 1) (Gottsberger and Silberbauer-Gottsberger 1988) suggesting pollination in this way would be unusual and it appears *S. obtusifolia* is self-fertile (Retzinger 1984). Indeed, self-fertilisation is probably normal in *S. obtusifolia* as the flower is commonly fertilised in late bud, before the flower is open, when the style is curved inward to present the stigmatic cavity directly to the face of the precociously dehiscent anthers (Irwin and Barneby 1982).

Sicklepod is a short-day plant. Short (12 h day) photoperiods accelerate reproductive development in sicklepod; a 15 h day delays the appearance of buds and flowers and open flowers develop at higher nodes than in plants reared in short day lengths (Patterson 1992). Patterson (1993) showed that anthesis occurred at 34+/-1 day from germination at 12 h light and 46+/-2 days at 14 h light, whilst at 16 h light, plants failed to flower. This response varied with ecotype as plants from a different ecotype had a threshold of 15 h light for normal reproductive development. Exposure to at least two weeks of short days is required to initiate reproductive development. With circa 2 weeks exposure, flower buds appear in 20-21 days, flowers after 30-40 days and pods after 35-43 days. For post bud development, a longer exposure to short days is required: at around 6 weeks of exposure 95-100% of plants produced flowers and 85-95% produced pods.

6.5 Seed

A single sicklepod plant can produce up to 8000 seeds (Retzinger 1984, Hall and Vandiver 1996). Studies in the USA have shown that the number of seeds per pod depends on ecotype and the climate (year) and varied from 24-28 seeds per pod. The number of pods per plant also varies greatly, between 63 and 342. The average number of seed per plant varies from 5280-8520 for the highest yielding ecotypes with an average of 1500-1600 overall (Retzinger 1984). Such high production leads to high seed yields: up to 3000 kg/ha are recorded (Anning *et al*. 1989, Anon. 1989) and to large soil seed banks (300 kg ha^{-1}). The number of seeds per pod is less in foetid cassia than sicklepod (Singh 1968). Seeds are scattered from the pods as the pods dry and open (Anning *et al*. 1989).

The seed coat is hard, giving protection of up to five years in the soil (Anning *et al*. 1989) or even 10 years (Anon. 1989) and an anecdotal account suggests *S. tora* seed may be viable for 20 years or more (Cock and Evans 1984). The seed is covered with a wax-like layer 0.1 mm thick and the embryo is surrounded by a dense hard material (Creel *et al*. 1968). The majority of seed of both species requires scarification to germinate (Creel *et al.* 1968, Singh 1968, Teem *et al.* 1980): in field trials, germination of non-scarified sicklepod seed varied from 1.6%-11% with an average of 4.5% in 1981 and 3%-23% in 1982 (Retzinger 1984). Unscarified seed incubated for 12 months in the soil gave only 15% germination, but most of these germinated in the first 30 days of the trial (Creel *et al*. 1968, Hall and Vandiver 1996). Untreated seed of foetid cassia species has negligible germination (2%) but acid scarified seed gave almost total germination (Cock and Evans 1984).

Fire scarifies the seed and in Australia has caused mass emergences of seedlings following good rain (Anning *et al*. 1989). Nevertheless, sicklepod is relatively resistant to drought as under simulated drought conditions a significant proportion of seed could germinate (Hoveland and Buchanan 1973). This capacity to germinate at relatively low soil moistures, combined with the high seed production may account for the rapid establishment of sicklepod and may explain why it can colonise bare soils and why it may have a particular advantage on sandy soils which dry out quickly (Hoveland and Buchanan 1973). Sicklepod seeds can germinate from a depth of 12.7 cm in the soil and this gives sicklepod a competitive advantage over many other plants (Teem *et al*. 1980). The seed germinates between 18 and 36^{o} C with an optimum range of 24-33^{o} C. The optimum temperature for seedling growth is 33^{o} C with growth occurring from 18-39^{o} C, but the optimum range is narrow (Creel *et al*. 1968).

After storage for one year, only about 50% of *S. obtusifolia* seed germinated, irrespective of storage temperature. *S. tora* seed germination after a year varied between 8 and 50%: the lower the storage temperature the lower the germination rate. This difference suggests *S. tora* should favour warmer climates and also supports the contention (Irwin and Turner 1960) that *S. tora* is derived from *obtusifolia* (Singh, J. S. 1968). It also supports the results of the modelling which suggest the potential distribution of *S. tora* in Australia is more restricted than that of *S. obtusifolia* and constrained to warmer areas.

Aqueous extracts of sicklepod can severely inhibit the germination of other plant seed depending on species and it is argued that sicklepod contains a non-persistent phytotoxic substance (Creel *et al*. 1968). This may explain the observation (Anon. 1989) that the growth of watermelons and pumpkins is reduced when grown in land previously infested by sicklepod.

6.6 Dispersal

Sicklepod and foetid cassia have a dehiscent pod which can disperse seed up to 5 m from the plant. Some seed does remain in the pod after dehiscence and this drops close to the base of the plant. Long distance seed dispersal in nature is mostly by stream flow, water movements over the soil surface or in mud attached to the feet and fur of animals. In weedy situations seed can be moved in mulch, in mud on machinery and vehicles and on footwear. Cattle, horses (Anning *et al*. 1989) and goats (Lock 1996 pers. comm.) will nibble the pods of sicklepod and ingest the seed, some of which survive passage through the gut and are spread in the dung (Anning *et* al. 1989, Anon. 1989). Stock are not known to browse on foetid cassia (Lock 1996 pers. comm.) and although it has been reported that stock will eat the plant in green silage and also the dry pods (Waterhouse and Norris 1987) because of the confusion over the identification of the two species, this must be considered a doubtful observation.

In sugar cane areas, dispersal is mainly by floodwaters, cane bins via the usually extensive tram line systems found in cane areas and harvesters. In the Herbert River area, grazing Magpie geese are thought to be dispersal agents, although this is unlikely (R. Wilson, *pers. comm.* 1997) and in this respect it is interesting to note that sicklepod seed is harmful to chickens due to the presence of a trypsin inhibitor (Cock and Evans 1984).

7.0 Efficacy of Current Control Methods

Effective control methods are not generally available for sicklepod and foetid cassia and research at the Tropical Weeds Research Centre is aimed at improving the efficiency of currently available techniques. Methods for their control in cropping and pasture situations can be can be expected to be different and have different efficacies.

7.1 Prevention

One of the main ways that sicklepod is spread is on machinery and vehicles and consequently good farm hygiene is the key to preventing new infestations. Wash down sites should be designated and should be monitored for emerging seedlings which should be treated immediately. Stock should be prevented from feeding on the pods and stock movements in infested areas should be restricted during podding.

7.2 Chemical Control

Sicklepod and foetid cassia are not amenable to chemical control except as seedlings (P. van Haaren *pers. comm.* 1996) or young plants up to 30 cm tall (L. Warren *pers. comm* 1997). Anecdotal information suggest that sicklepod growing in different areas on the Northern Territory and Queensland react differently to chemical control, perhaps depending on the woodiness of the growth from. The cuticle of the adult plant is thick and provides protection from herbicides (Anning *et al.* 1989): up to a 15-fold reduction in herbicide efficiency when plants are in the podding stage compared to the seedling stage (P. van Haaren *pers. comm.* 1996). Perhaps because of this, chemical control has often failed but testing has shown that control during the vegetative phase is possible with hexazinone (2 kh/ha), Tordon 50-D (1:200 in water) and dichloprop (0.5% in water) (James and Fossett 1982). Phenoxy herbicides such as 2,4-D amine 20/20 give good results on seedlings younger than 8 weeks but older plants only respond to Tordon. Herbicides should therefore be applied when plants are small (<30 cm) and actively growing at the start of the summer season (Anon 1989). In the Northern Territory, sicklepod does not respond well to any chemicals, including Tordon, as it seems to grow like a woody shrub (Schultze *pers. comm*.). Research at the Tropical Weeds Research Centre (P. van Haaren *pers. comm.* 1996) has shown that in non-crop situations high volume overall applications of Tordon 75-D (picloram) and Grazon DS (picloram and triclopyr) to the point of run-off is more effective than boom spraying (75-100% control). For effective control both methods require the addition of a non-ionic wetting agent. Suggested application rates are given in Table 3. Sicklepod appears to respond to control treatments in different areas, in different ways, so before any control is undertaken, advice should be obtained from local representatives of NR&M, DPI or BSES.

Pre-emergence control using atrazine, Gesapax Combi@ or Velpar K4@ applied to cane blocks at the out-of-hand stage, achieve some control (Anon 1989) but the standard treatment for cane blocks is to spray with Tordon 750@. In Florida (USA) 2,4- D amine is used to control sicklepod in corn at rates of 0.56 kg/ha (Anon 1996a) and control in crops often requires two herbicide applications (Anon. 1996c). The timing of herbicide application has been found to be critical in the USA as well as in Australia: best results are obtained when plants are 2.5-7.5 cm tall and have only 1 or 2 true leaves. Herbicides used in crops in the USA are imazaquin, flumetsulam, metribuzin and metribuzin + 2,4-0. Vencill *et al.* (1995) applied a pre-emergence broadcast of metribuzin when sicklepod was at the 1-2 leaf stage and about 3-7 cm tall. This, followed by either one or two cultivations gave about 90% control of sicklepod and this was about as effective as pre-emergence broadcast of metribuzin followed by a post- emergence dose of 2,4-0 or chlorimuron. Trifluralin usually provides poor control of sickle pod (Vencill *et al.* 1995).

Table 3. Suggested herbicide application rates for the control of sicklepod in grazing and cane growing areas.

Herbicide	**Method**	**Rate**	**Comments**
Grazing*			
Tordon 75-D ®	High volume overall spray	1:300	early seedling vegetative stage
		1:150	early flowering stage
		1:75	podding stage
Grazon DS ®		1:500	prior to onset of flowering
Tordon 75-D ®	Boom	670 mL ha^{-1}	seedling vegetative stage
		1.3 L ha^{-1}	early flowering stage
		2 L ha^{-1}	after seed set

Cane Ψ			
Tordon 75-D ® + 2,4-D	Boom	0.7 L + 1.0 L (500 g L^{-1})	height <0.5 m
		1.0 L + 1.0 L (500 g L^{-1})	height 0.5 - 1.0 m
		1.5 L + 1.0 L (500 g L^{-1})	height >1.0 m

* Use non-ionic wetting agent

Ψ Control is improved if a spraying oil is added. **A 150 day withholding period is mandatory.**

Maintaining sicklepod populations at economic threshold levels in crops results in large increases in the sicklepod seed bank (Anon 1996c) so almost complete control and prevention of seeding over a number of years is required for adequate sicklepod control. The fact that sicklepod seed can germinate in depths of soil to 12.7 cm gives the plant a good mechanism for escaping most soil applied herbicides (Teem et *a/.* 1980).

Sicklepod shows marked endogenous, circadian leaf movements which have been studied by Kraatz & Andersen (1985). These movements significantly alter the projected leaf area of the plant. Projected leaf area is greatest in early-mid morning and mid-late afternoon (about 2.5 h after sunrise and before sunset). These peaks occur irrespective of weather and sky conditions. The volume of spray retained by a plant is directly related to the projected leaf area and in trials, based on shoot dry weight reductions, the control of sickle pod seedlings was strongly correlated with projected leaf area. Diurnal movement of leaves could therefore be a significant factor in achieving sicklepod control by herbicides and should be taken into account when planning control measures.

7.3 Mechanical control

Slashing plants in their vegetative phase reduces their size and delays seeding but does not kill the plant, though if plants in the flowering or early podding stages are slashed with blunt blades to shatter the stems, a 'fair' percentage kill is achieved (Anning *et al.* 1989, Anon. 1989, James and Fossett 1982, P. van Haaren *pers. comm.* 1996). Opinion is divided on the utility of slashing as a control device: it is impractical on hillsides and whilst it can prevent increases in the seed bank, reduce weed vigour, and encourage the growth of native pasture, often it is not considered a successful control in pasture areas, as it causes the plant to perenniate (James and Fossett 1982). Repeated slashing over a number of years may cause the weed to become procumbent, in which state it can still set seed (J. Arnold, *pers. comm.* -1997). Conversely, some graziers with heavy infestations of sicklepod on parts of their properties, consider slashing to be worthwhile. Slashing plants just prior to flowering, sometimes followed by spraying of the regrowth, is considered to be an effective way of stopping seed production, limiting dispersal and depleting the seed bank. Ploughing is not successful and usually causes an increase in plant numbers (James and Fossett 1982) due to the scarification of seeds in the soil during the ploughing process.

7.4 Fire

Fire does not seem to have been evaluated as a control for sicklepod and foetid senna in grazing systems. However, as noted previously, fire scarifies the seed and in Australia has caused mass emergences of seedlings following good rain (Anning *et al.* 1989).

In natural areas which are strategically burnt as part of a management plan, fire may be a useful tool to control sicklepod invasion: In Dryander National Park, the open forest areas are heavily infested by sicklepod. Strategic burns over these areas were initially carried out every 2 years, but are now burnt on a 3-5 year cycle, and are considered to be a valuable tool for controlling sicklepod invasion by killing seedlings (B. Nolan, *pers. comm.* 1997).

No information is available on the effects of pre-harvest cane burning on sicklepod infestations in cane areas.

7.5 Biological Control

It may be possible to use exotic insects or pathogens in a classical biological control programme for sicklepod and foetid cassia. In this methodology the exotic agent is imported, tested to ensure safety and then released throughout the range of the weed. It is then expected to build up to damagingly high population densities, spread, and reach a population equilibrium, with only minimal human manipulation.

Because of the difficulty of controlling sicklepod, particularly in crops, control research has emphasised biological control, and particularly the use of pathogenic fungi as mycoherbicides. Mycoherbicides have the advantage over conventional herbicides in that they use native fungal species rather than exotics, are species specific and do not pose a problem to non-target species (Auld 1990). The development and use of a fungal pathogen as a mycoherbicide compensates for its (usually) poor natural dispersal ability and relies on a high level of target specificity and its strong necrotrophic ability at elevated inoculum levels. Such species are generally constrained from reaching epidemic development by their innate deficiency to disseminate. They usually infect and kill the plant within 3-5 weeks and after the death of the host, the pathogen is reduced to background levels by natural constraints and deterioration (Templeton 1987). The fungus *Alternaria cassiae* Jurair and Khan is pathogenic to seedlings of sicklepod and coffee senna (*S. occidentalis* L.) but not to 30 major crop species (Walker 1982) and it also attacks foetid cassia (Cock and Evans 1984). Walker and Riley (1982) showed that seedlings in the cotyledon to first leaf stage are most sensitive to the pathogen and that there is a dramatic increase in tolerance as plants increase in size. At the fist leaf stage, 96% of plants were killed, but at the second and third leaf stages only 15-18% of plants were killed, although surviving plants were severely stunted (~75% reduction in dry weight). Plants in the fourth leaf stage were not killed and only evinced a 25% reduction in dry weight. Inoculua of 3-5 x 10^4 conidia mL^{-1} gave maximum control at the optimal environmental conditions of 8 h of free moisture at 20-30 ^oC (Walker and Riley 1982). To kill infected seedlings, *A. cassiae* requires 8 h of dew within 2 days of inoculation and to be used as a microbial herbicide, inoculum has to be applied when dew is likely to form on the foliage (Walker and Boyette 1986). Field trials showed that *A. cassiae* killed 90-94% of plants with a second field application increasing control to 98% (Boyette 1988).

The use of *A. cassiae* as a mycoherbicide was considered very promising for many years and this has possibly restricted the search for more conventional biocontrol agents of sicklepod. The mycoherbicide was planned for release in 1990 to be used at the rate of 1.1 Kg ha^{-1} in 76.6 L of water, with an oil based adjuvant and it was expected that it could be used in conjunction with chemical herbicides (Auld 1990). However the product was never released and further research and development by the company concerned was shelved for the following reasons (S. Savage 1996 pers. comm.):

- *A. cassiae* was very difficult to produce on a large scale - it did not sporulate in deep tank fermentations and mycelial inoculum was also non-economic.

- Infection requires a dew period (Walker and Riley 1982). This limitation is critical. Although research had indicated that *A. cassiae* could infect sicklepod within 4 hours of a dew, this infection rate only occurs at temperatures which are quite a bit higher than those common in the field. Previous research had been unrealistically optimistic in relation to this requirement. A great deal of research was aimed at overcoming this limitation, but nothing practical ever came of these efforts.

- *A. cassiae* is really only effective against very small seedlings of sicklepod. Since the emergence of the weed from the seed bank is not uniform, either some plants would be too large to kill or would not have emerged at the time of the treatment.

- *A. cassiae* is very selective for sicklepod. Initially this was acceptable but in a cropping situation, a more broad spectrum approach is required to cope with the other weeds present.

Several other species of pathogenic fungi have been considered as candidate mycoherbicides for the control of sicklepod: *Fusarium oxysporum* Schlet. emend Synd. and Hans. (Boyette *et al*. 1993), *Pseudocercospora nigircans* (Cooke) Deighton (Hofmeister and Charudattan 1987), *Coletotrichum fragariae* Brooks (Howard and Albregts 1973), *Pseudocercospora nigricans* Cooke Deighton, *Pseudopeonospora cassiae* Waterhouse & Brothers and *Ravenelia berkeleyii* Mundkur & Thirumalachar (Cock and Evans 1984). Although as late as 1993, Boyette *et al.* were suggesting that as *F. oxysporum* was soil borne and had the advantage over air borne pathogens of being buffered from adverse environmental conditions, it had good potential as a mycoherbicide it is unlikely that this or any of the other species will be developed further as the limitations on production outlined in the first point above, are common to most, although not all, mycoherbicides. Further developments in the biocontrol of sicklepod will, at least in the short to medium term, depend on finding more conventional insect biocontrol agents.

Table 4. Natural enemies recorded from sicklepod (*Senna obtusifolia*) and foetid cassia (*S. tora*) after (Cock and Evans 1984) and (Waterhouse and Norris 1987).

Order: Family	**Species**	**Locality**	**Host**
Hemiptera: Alydidae	*Leptocorisa acuta*	Fiji	*S. tora*
Coleoptera: Bruchidae	*Callosobruchus chinensis* L.	India	*S. tora*
	Bruchidius cassiae Arora	India	*S. tora*
	Caryedon lineatonota Arora	India	*S. tora*
	Amblycerus sp. 2	Costa Rica	*S. obtusifolia*
	Caryedon pallidus (Olivier)	Nigeria	*S. obtusifolia*
	Sennius fallax (Boheman)	USA: Florida, Georgia	*S. obtusifolia*
	S. instabilis (Sharp)	Costa Rica, Mexico, Trinidad	*S. obtusifolia*
	S. morosus (Sharp)	Costa Rica	*S. obtusifolia*
	S. rufescens (Motschulsky)	Mexico	*S. obtusifolia*
Curculionidae	*Hypocoeliodes* sp. *?nebulosus* Hustache	Trinidad	*S. obtusifolia*
	Perigaster sp. *?tetracantha* Champion	Trinidad	*S. obtusifolia*
	Elytrurus griseus	Fiji	*S. tora*
	Zena sp. *?virgata* (Boheman)	Philippines	*S. tora*
Chrysomelidae	*Pseudoides flavovittis* (Motschulsky)	Thailand	*S. tora*
Lepidoptera: Pieridae	*Eurema* sp.	Trinidad	*S. obtusifolia*
	Catopsilia scylla	Papua New Guinea	*S. tora*
	Eurema hecabe	Fiji	*S. tora*
	Phoebis sennae (L.)	Trinidad	*S. obtusifolia*
Hesperidae	*Typhedanus undulatus* Hewitson	Trinidad	*S. obtusifolia*
Gelechiidae	*Stegasta variana* Meyr.	Vanuatu	*S. tora*
Pyralidae	Phycitinae indet. sp.	Trinidad	*S. obtusifolia*
	Anabasis ochrodesma	USA, Mexico, Trinidad, Colombia	*S. obtusifolia*
Diptera: Agromyzidae	*Calcomyza* sp.	Trinidad	*S. obtusifolia*

The prospects for the biocontrol of sicklepod and foetid cassia by insects have been reviewed by Cock and Evans (1984) and Waterhouse and Norris (1987). Table 2 lists the natural enemies so far collected from these two plant species. The principal natural enemies are found amongst the oligophagous, seed feeding Bruchidae: three species have been found on *S. tora* and six species on *S. obtusifolia*. In addition eight species of

Lepidoptera, five other species of Coleoptera, one species of Hemiptera and one species of Diptera are recorded from these weeds. Doubtless many other natural enemies exist and further field surveys should be carried out. Cock and Evans (1984) and Palmer (unpublished) consider that biocontrol is more likely to be successful for these sicklepod and foetid cassia in a pasture situation. For the control of *S. tora* in the Old World, the seed bruchid *Sennius instabilis* which attacks *S. obtusifolia* in the New World, should be considered (Cock and Evans 1984) although the authors recognise that because of the high seed production, the effects of a seed predator such as this may not be effective on the infestation until the seed bank had been significantly depleted. However, a seed predator might slow the rate at which new infestations are established (Waterhouse and Norris 1987).

Cock and Evans (1984) suggest that natural enemies may be interchangeable between the two *Senna* species because of their close taxonomic affinity but Waterhouse and Norris (1987) note that as yet, there is no evidence to suggest that an insect host-specific to sicklepod will attack foetid cassia or be of use in its control and that in areas where the distributions of the two weeds overlap, the only insects which feed on both are two widely polyphagous pest species.

Since the reviews of Cock and Evans (1984) and Waterhouse and Norris (1987) a survey of the phytophagous insects associated with *S. obtusifolia* in Mexico has been carried out between 1992 and 1994 by W. A. Palmer for the Department of Natural Resources and Mines (Land Protection). The plant was found to be distributed along both the Gulf and Pacific coasts of that country and across the Yucatan peninsula. It was also found to be abundant in Honduras although no formal study was undertaken there.

Table 5. Phytophagous insects collected on *S. obtusifolia* that are considered to be potential biocontrol agents.

Coleoptera
Bruchidae
Sennius fallax (Boheman)
Sennius lebasi (Fahraeus)
Sennius morosus (Sharp)
Curculionidae
Conotrachelus sp.
Homoptera
Psyllidae
Mitrapsylla albalineata Crawford
Lepidoptera
Hesperiidae
Typhedanus undulatus (Hewitson)
Pieridae
Eurema albula Cramer
Pyralidae
Anabasis ochrodesma (Zeller)

Some 105 insect species were collected on *S. obtusifolia* in Mexico. Although no formal host range testing has been undertaken some species have been selected as probably having a narrow host range (Table 4). Potential biocontrol agents would come from this group. A Brazilian group led by Dr. E. Fontes has been investigating biological control for this weed. They suggest four possible agents (Fontes *et al.* 1995, Sujii *et al.* 1996): *Agrilus oceanicum* (Buprestidae), *Fundella argentina* (Pyralidae), *Thyphedanus undulatus* (Hesperidae) and *Phoebis sennae* (Pieridae). It is doubtful if these species would be sufficiently stenophagous for use in Australia.

One unanswered question of some practical relevance is whether insects associated with *S. obtusifolia* will also attack the Old World species *S. tora*. As these species are very similar morphologically and are phylogenetically close, it is likely that insects will attack both species (Cock and Evans 1984, Palmer unpublished).

There has been no systematic search for the phytophagous fauna of *S. tora* in its natural habitat of Africa and Asia although a few species are listed(Cock and Evans 1984, Waterhouse and Norris 1987). The fauna of this plant might well produce biocontrol agents for both weeds.

One factor which will limit the number of available biocontrol agents is the high degree of host specificity that will be required. Both *S. obtusifolia* and *S. tora* are placed in section *Chamaefistula* of the genus *Senna*. There are three endemic Australian species in this section (Appendix A) so that any biocontrol agent will need to be very close to monophagous to be approved for release. However, of the three species, *S. barclayana* is a weed of cultivation and pasture, *S. plantiicola* seems to be basically a western species with its range only marginally coinciding with *S. obtusifolia* and whilst the distribution of *S. clavigera* more closely coincides with that of *S. obtusifolia*, as it is found along the eastern coast from Charters Towers to southern NSW and is probably the species of greatest concern, it may also be weedy. Thus, for varying reasons of abundance, geographic distribution and weediness, it is quite possible that insects that also attacked any of these three natives in the section *Chamaefistula* would be acceptable.

Sicklepod and foetid cassia produce a significant seed bank in the soil and in no tillage systems, sicklepod seeds are preferentially fed upon by soil arthropods, primarily by carabid beetles in the omnivorous genus *Harpalus* (Brust & House 1988). Consumption of sicklepod seed was three times more in no tillage systems than in conventional tillage systems, which tended to have a depleted soil fauna. These observations may be of significance to biocontrol in grazing systems in Australia where sicklepod is a weed, and soils arthropods have not been reduced by tilling and insecticide application. However, Harpalini are not as well represented in Australia as elsewhere in the world (CSIRO 1991).

8.0 Management and Control Practices

8.1 Legislative Status in Queensland

Sicklepod is a declared plant under the provisions of the *Rural Lands Protection Act* and for the purposes of this Act also includes foetid senna. The species are declared in both Categories P3 and P4 in the local government areas of Bowen, Burdekin, Cairns, Cardwell, Cook, Douglas, Hinchinbrook, Johnstone, Mackay, Mulgrave, Pioneer, Proserpine, Sarina, Thuringowa and Townsville. All P3 infestations are to be reduced in area and P4 infestations should be prevented from spreading. The species are declared as P2 for the remainder of the state and the plant must be destroyed.

8.2 Containment Strategies in Queensland

Sicklepod is well established in Queensland. Its continued spread requires containment and the areas of current infestation need to be gradually reduced. Some intractable core infestations are recognised as providing source material for reinvasions of controlled areas and the control of these infestations needs to be addressed in consultation with the individuals and communities involved.

The ecoclimatic models for both sicklepod and foetid cassia suggest that the species could invade the southern parts of Queensland and that foetid cassia could invade coastal areas around the Gulf of Carpentaria. These dispersal movements may be guarded against by monitoring for new infestations in these areas and treating them as soon as they are discovered. Mapping the extent of the current infestations of sicklepod would be invaluable in this regard, an indeed, some northern shires are carrying out such mapping as part of their weed management strategies.

Spread can be prevented by careful management of stock movement when the plant is podding and seed is being released, and by washing down equipment after it has been used in infested areas. Currently, chemical control is the only viable method of containing and reducing the sicklepod problem. Since sicklepod becomes less amenable to chemical control as it grows older, the timing of herbicide application is critical to its successful control and whenever possible, control should be initiated in the seedling stage. Landholders in infested areas need to be fully aware of this when carrying out control activities. The seed is dispersed by water movements, so upstream infestations in a catchment should be brought under control first. In cane growing areas, the spread of the weed by the tramway system should be guarded against. In these areas, tram ways and water ways should be patrolled during the growing season and any plants found spot sprayed to ensure control before the plants seed. Because the weed grows in inaccessible areas and because of the extensive, and in many areas very dense, nature of the current infestations, the future containment of the spread of sicklepod may rely on finding suitable biocontrol agents for sicklepod.

8.3 Eradication Strategies in Queensland

Both sicklepod and foetid cassia are well established in Queensland and state wide eradication is therefore not feasible. New infestations can be guarded against and it may be possible for these to be eradicated if treated sufficiently early and follow up treatments in subsequent years are undertaken until the seed bank is exhausted.

Sicklepod has the potential to become a major weed across much of Queensland and although its current range is extensive, the weed is only in the early stages of its invasion. The economic impact of this weed could become significant and it may therefore be a good candidate for a major, coordinated control programme across the state, although the narrow window of opportunity for control present in the sicklepod life cycle, and its annual nature, may mitigate against this.

8.4 Property Management Strategies

Neither species is considered to be amenable to chemical or cultural control (Cock and Evans 1984) and no biocontrol agents are available. Control methods must therefore rely heavily upon property management practices. These will be different in pasture and cropping areas.

Sicklepod is not usually a pest in well managed tropical and native pastures so the initial management practice is to keep pastures in good condition (James and Fossett 1982). In grazing areas Anning *et al.* (1989) suggest that the key to control is a vigorous pasture. Sicklepod does not occur in vigorous, sown tropical pastures with a dense ground cover. However, once present, sicklepod can colonise bare areas in a pasture. It then thickens, increasing grazing pressure on the remaining pasture and takes over. In infested areas, animals should be kept out of blocks having plants with mature seed heads and the production of viable seed should be eliminated or reduced by slashing or chemical control (Anon. 1989).

A sicklepod infested area should be cultivated and sown to grass on a moist soil and rolled. Grasses used in the past are signal grass (*Brachiaria decumbens* Stapf), humidicola (*B. humidicola* (Rendle) Schwenke), pangola (*Digitaria decumbens* Stent) and gamba grass (*Andropogon gayanus* Knuth) (Anning 1989) but the latter three species are considered as weeds or problem plants in some countries overseas and presently signal grass and Kazungula setaria (*Setaria sphacelata* cv. *kazungula*) are considered the most suitable for sowing. Tordon intolerant grasses should not be used as Tordon is the only effective herbicide for spraying the dense sicklepod seedling population which will develop. This should be sprayed whilst the seedlings are young and certainly before they flower. A second spray may be required a month later. To ensure a vigorous pasture, fertilise with 100 kg ha^{-1} y^{-1} DAP every two years and split applications of urea in February and April at 100-200 kg ha^{-1} y^{-1}. Where cultivation is not possible on hillsides, runners can be planted by hand on a 3 x 3 m grid. Good rain is required to enable the grass to colonise adequately (Anning *et al*. 1989). In situations where planting introduced grasses is not suitable, pasture spelling and perhaps burning may be viable control options.

In cultivated areas, ploughing by itself is not a successful method of control and usually causes an increase in sicklepod density (James and Fossett 1982). However, in soybeans the use of a pre-emergence herbicide followed by two cultivations achieved far better control of sicklepod than herbicide alone (Smith *et al.* 1986) and this practice may have application in sugar cane areas. In cane growing areas the weed should be controlled on headlands around cane assignments. It should be prevented from flowering and follow up treatments should be made until the seed bank is exhausted.

9.0 References

Abaye, O. 1992. Sicklepod, a weed to be avoided. URL:http://www.ext.vt.edu/docdb/cses/cses-267 (19/03/96).

Anning, P., H. G. Bishop, G. Lambert, and G. Sutherland. 1989. Sicklepod replaces overgrazed tropical pastures. Queensland Agricultural Journal **115**: 188-192.

Anon. 1989. Sicklepod - another problem weed for the tropics. BSES Bulletin (No. 27): p.18-19.

---. 1996a. Chemical weed control in corn. URL:http://hammock.ifas.ufl.edu/txt/fairs/wg/3.html (19/03/96).

---. 1996b. Sicklepod (*Cassia obtusifolia*). Weed Control Guide (WG153) (Florida). URL:http://hammock.ifas.ufl.edu/txt/fairs/ (19/03/96).

---. 1996c. Sicklepod control. URL: http://ipmwww.ncsu.edu/soybeans/weeds/weedmanage/weedmanagement8.html.

Auld, B. A. 1990. Mycoherbicides: new tools in weed management. BIOTROP Special Publication (No. 38). pp.11-15.

Boyette, C. D. 1988. Biocontrol of three leguminous weed species with *Alternaria cassiae*. Weed Technology **2**: 414-417.

Boyette, C. D., H. K. Abbas, and W. J. Connick. 1993. Evaluation of *Fusarium oxysporum* as a potential bioherbicide for sicklepod (*Cassia obtusifolia*), coffee senna (*C. occidentalis*), and hemp sesbania (*Sesbania exaltata*). Weed Science **41**: 678-681.

Brenan, J. M. 1958. New and noteworthy Cassias from tropical Africa. Kew Bulletin **13**: 231-252.

Brust, G.E. and House, G.J. 1988. Weed seed destruction by arthropods and rodents in low-input soybean agroecosystems. American Journal of Alternative Agriculture **3**: 19-25.

Buchanan, G. A., and C. S. Hoveland. 1973. Soil phosphorus and potassium levels on growth of warm season weed and crop species. Abstracts, 1973 Meeting of the Weed Science Society of America, Atlanta, Georgia.p.53.

Buchanan, G. A., C. Hoveland, and M. C. Harris. 1975. Response of weeds to soil pH. Weed Science **23**: 473-477.

Buchanan, G. A., R. H. Crowley, and J. A. McGuire. 1978. Economic thresholds of sicklepod (*Cassia obtusifolia* L.) in cotton. Weed Science Society of America Abstracts. p. 65.

Cock, M. J. W., and H. C. Evans. 1984. Possibilities for biological control of *Cassia tora* and *Cassia obtusifolia*. Tropical Pest Management **30**: 339-350.

Creel, J. M., C. S. Hoveland, and G. A. Buchanan. 1968. Germination, growth and ecology of sicklepod. Weed Science **16**: 396-400.

CSIRO, 1991. The Insects of Australia: a textbook for students and research workers. (2nd. Edn.). Melbourne University Press. Carlton, Victoria.

Currey, W.L., D.H. Teem, and J.H. Jordan. 1981. Sicklepod competition and control programs in Florida soybeans. p.66. Proceedings of the 34th Annual Meeting, Southern Weed Science Society, Florida University, Gainesville, FL 32611, USA.

Dowling, R. M., and R. A. McKenzie. 1993. Poisonous Plants: a field guide. Department of Primary Industries, Brisbane.

Farnsworth, E. J., and F. A. Bazzaz. 1995. Inter- and intra- generic differences in growth, reproduction and fitness of nine herbaceous annual species grown in elevated CO_2 environments. Oecologia (Berlin) **104**: 454-466.

Flint, E. P., D. T. Patterson, D. A. Morteson, G. H. Reichers, and J. L. Beyers. 1984. Temperature effects on the growth and leaf production in three weed species. Weed Science **32**: 655-663.

Fontes, E. G., C. S. S. Teixeira, and E. R. Sujii. 1995. Current status of the biological control of weeds in Brazil. Proceedings of the VIII International Symposium on Biological Control of WEEDS. 2-7 February 1992, Lincoln University, Canterbury, New Zealand. E. S. Delfosse and R. R. Scott (ed.), DSIR/CSIRO, Melbourne. 693 pp.

Gottsberger, G., and I. Silberbauer-Gottsberger. 1988. Evolution of flower structures and pollination in neotropical Cassiinae Caesalpiniaceae species. Phyton (Horn) **28**: 293-320.

Hall, D. W., and V. V. Vandiver. 1996. Sicklepod. URL: http: //hammock.ifas.ufl.edu/ txt/fairs/4924.

Hauser, E.W., G.A. Buchanan, R. Nichols, and R.M. Patterson. 1982. Effects of Florida beggarweed (*Desmodium tortuosum*) and sicklepod (*Cassia obtusifolia*) on peanut (*Arachis hypogaea*) yield. Weed Science **30**: 602-604.

Hofmeister, F. M., and R. Charudattan. 1987. *Pseudocercospora nigricans*, a pathogen of sicklepod (*Cassia obtusifolia*) with biocontrol potential. Plant Disease **71**: 44-46.

Hoveland, C. S., and G. A. Buchanan. 1973. Weed seed germination under simulated drought. Weed Science **21**: 322-324.

Howard, C. M., and E. E. Albregts. 1973. *Cassia obtusifolia*, a possible reservoir for inoculum of *Colletrotrichum fragariae*. Phytopathology **63**: 533-534.

Hutchinson, J., and J.M. Dalziel. 1937. The useful plants of west tropical Africa. London. pp. 612.

Irwin, H. S., and R. C. Barneby. 1982. The American Cassinae. A synoptical revision of Leguminosae tribe *Cassieae* subtribe *Cassiinae* in the New World. Memoirs of the New York Botanical Garden **35**: 252-255.

James, P. A., and G. W. Fossett. 1982/83. Sicklepod (*Cassia obtusifolia* L.) in Central Queensland. Australian Weeds **2**: 80-81.

Nicholson, S. S., W. Flory, and L. P. Ruhr. 1985/86. Sicklepod poisoning in cattle: a new development. Louisiana Agriculture **29**: 18-19.

Parsons, W. T., and E. G. Cuthbertson. 1992. Noxious Weeds of Australia. Inkata Press, Melbourne.

Patterson, D. T. 1992. Effects of photoperiod on vegetative growth and reproductive development of northern and southern biotypes of velevetleaf (*Abutilon theophrasti*) and sicklepod (*Cassia obtusifolia*). Weed Science Society of America Abstracts **32**: 53. (Abstract No. 159)

---. 1993. Effects of temperature and photoperiod on growth and development of Sicklepod (*Cassia obtusifolia*). Weed Science **41**: 574-582.

Quisumbing, E. 1951. Medicinal plants of the Philippines. Department of Agriculutre and Commerce, Philippine Islands Technical Bulletin **16**: 1-1234.

Randell, B. R. 1988. Revision of the Cassiinae in Australia. 1. *Senna* Miller sect. *Chamaefistula* (Colladon) Irwin and Barneby. Journal of the Adelaide Botanic Gardens **11**: 19-49.

---. 1995. Taxonomy and evolution of *Senna obtusifolia* and *S. tora*. Journal of the Adelaide Botanic Gardens **16**: 55-58.

Retzinger, E. J. 1984. Growth and development of sicklepod (*Cassia obtusifolia*) selections. Weed Science **32**: 608-611.

Singh, J. S. 1968. Comparison of growth performance and germination of seed oc *Cassia tora* L. and *C. obtusifolia* L. Tropical Ecology **9**: 64-71.

Singh, V. 1978. Critical taxonomic notes on some Indian species of *Cassia* Linn. found in India. Journal of the Bombay Natural History Society **75**: 434-443.

Skarratt, D. B., R. W. Sutherst, and G. F. Maywald. 1995. CLIMEX for Windows Version 1.0. User's Guide. CSIRO/CRC for Tropical Pest Management, Brisbane.

Smith, C.A., Shaw, D.R. and Snipes, C.E. 1986. Influence of cultivation on chemical control of sicklepod in soybeans. Proceedings of the 39th Annual Meeting, Southern Weed Science Society. p. 401.

Sujii, E.R., Fontes, E.G., Pires, C.S.S. and Teixeira, C.A.D. 1996. Application of multivariate analysis for the selection of candidates for biological control agents. Biological Control **7**: 288-292.

Symon, D. E. 1966. A revision of the genus *Cassia* L. (Caesalpinaceae) in Australia. Transactions of the Royal Society of South Australia **90**: 73-146.

Teem, D.H., C.S. Hoveland, and G.A. Buchanan. 1974. Primary root elongation of three weed species. Weed Science **22**: 47-50.

Teem, D. H., C. S. Hoveland, and G. A. Buchanan. 1980. Sicklepod (*Cassia obtusifolia*) and coffee senna (*Cassia occidentalis*) geographic distribution, germination and emergence. Weed Science **28**: 68-71.

Templeton, G. E. 1987. Mycoherbicides - achievements, developments and prospects. Proceedings of the Eighth Australian Weeds Conference. pp. 489-497.

Upadhyaya, S. K., and V. Singh. 1986. Phytochemical evaluation of *Cassia obtusifolia* L. and *Cassia tora* L. Proceedings of the Indian Academy of Science (Plant Science) **96**: 321-326.

Vencill, W. K., J. W. Wilcut, and C. D. Monks. 1995. Efficacy and economy of weed management systems for sicklepod (*Senna obtusifolia*) and morining glory (*Ipomoea* spp.) control in soybean (*Glycine max*). Weed Technology **9**: 456-461.

Walker, H. L. 1982. Seedling blight of sicklepod caused by *Alternaria cassiae*. Plant Disease **66**: 426-428.

Walker, H. L., and C. D. Boyette. 1986. Influence of sequential dew periods on biocontrol of sicklepod (*Cassia obtusifolia*) by *Alternaria cassiae*. Plant Disease **70**: 962-963.

Walker, H. L., and J. A. Riley. 1982. Evaluation of *Alternaria cassiae*) for the control of sicklepod (*Cassia obtusifolia*). Weed Science **30**: 651-654.

Waterhouse, D. F., and K. R. Norris. 1987. *Cassia tora* and *C. obtusifolia*. Pages 288-295 *in* Biological Control. Pacific Prospects. Inkata Press, Melbourne.

Whitt, D. 1992. Sicklepod leading problem in seed shipments. URL: http://www.ext.vt.edu/docdb/cses/cses-253 (19/03/96).

Appendix A

The Australian species of Cassiinae according to Dr. B. Randell

Australian Species of *Senna*

- Section *Chamaefistula*
  - Series *Basiglandulosae*
    - *S. barclayana* (Sweet) Randell
    - *S. plantiicola* (Domin) Randell
    - *S. clavigera* (Domin) Randell
    - *S. occidentalis* (L.) Link *
    - *S. hirsuta* (L.) Irwin and Barneby *
  - Series *Coluteoideae*
    - *S. bicapsularis* (L.) Roxb. *
    - *S. corymbosa* (Lam.) Irwin and Barneby *
    - *S. x floribunda* (Cav.) Irwin and Barneby *
    - *S. multiglandulosa* (Jacq.) Irwin and Barneby *
    - *S. pendula* (Willd.) Irwin and Barneby *
  - Series *Trigonelloideae*
    - *S. obtusifolia* (L.) Irwin and Barneby *
    - *S. tora* (L.) Roxb. *

- Section *Psilorhegma*
  - Series *Interglandulosae*
    - *S. acclinis* (F. Muell.) Randell
    - *S. aciphylla* (Benth.) Randell
    - *S. coronilloide* (Benth.) Randell*s*
    - *S. costata* (J. F. Bailey and C. White) Randell
    - *S. odorata* (Morris) Randell
    - *S. surattensis*(Burman f.) Irwin and Barneby
  - Series *Subverrucosae*
    - *S. artemisoides* (DC.) Randell
    - *S. cardiosperma* (F. Muell.) Randell
    - *S. glutinosa* (DC.) Randell
  - Series *Oligocladae*
    - *S. procumbens* Randell
    - *S. cladophylla* (W. Fitzg.) Randell
    - *S. curvistyla* (J. Black) Randell
    - *S. goniodes* (A. Cunn. ex Benth.) Randell
    - *S. heptanthera* (F. Muell.) Randell
    - *S. leptoclada* (Benth.) Randell

- *S. oligoclada* (F. Muell.) Randell

Section *Senna*

- Series *Pictae*
    - *S. venusta* (F. Muell.) Randell
    - *S. notabilis* (F. Muell.) Randell
    - *S. magnifolia* (F. Muell.) Randell
    - *S. pleurocarpa* (F. Muell.) Randell
    - *S. didymobotrya* (Fresen.) Irwin and Barneby *
    - *S. alata* (L.) Roxb. *
- Series *Floridae*
    - *S. timoriensis* (DC.) Irwin and Barneby

- introduced species

Australian Species of *Cassia*

C. auriculata L.†
C. brewsteri ((F. Muell.) F. Muell. ex Benth.
C. concinna Benth.†
C. fistula (L.) *
C. grandis L. f. *
C. harneyi Specht†
C. longipes Domin†
C. marksiana L.
C. queenslandica C. White
C. tomentella Domin

† these species should be moved to *Chamaecrista* (Randell, pers. comm.)

Australian Species of *Chamaecrista*

C. absus L.
C. mimosoides L.
C. pumila Lam.†
C. rotundifolia Pers.

* introduced species

Pest Status Reviews

Others in the series:

Prickly acacia in Queensland

Rubber vine in Queensland

Mesquite in Queensland

Cabomba in Queensland

Goats in Queensland

Veterbrate Pests of Built-up areas

House mouse in Queensland

Bellyache bush in Queensland

Feral pigs in Queensland

Hymenachne in Queensland

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