Chlorophyll Biosynthesis

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INTRODUCTION

A leaf with 70 million cells houses \( \sim 5 \times 10^9 \) chloroplasts, each containing \( \sim 600 \) million molecules of chlorophyll (Simpson and Knoetzel, 1995). These \( 10^{18} \) chlorophyll molecules, all of which are bound to proteins of the photosynthetic membranes, harvest the sunlight. Approximately 250 to 300 of them transfer the absorbed light energy through neighboring pigments to the "special pair" chlorophylls in a reaction center. These special pair chlorophylls in photosystems I and II are the primary electron donors that drive the conversion of light into chemical energy to be conserved in NADPH2 and ATP.

In chlorophyll, four pyrrole rings (designated I to IV) are ligated into a tetrapyrrole ring with a magnesium atom in the center (Figure 1). Ring IV is esterified with phytol. Chlorophyll a has a methyl group in position 3, but higher plants and algae use for light harvesting an additional form of chlorophyll, chlorophyll b, that has a formyl group instead of a methyl group at this position. The porphyrin ring with its conjugated double bonds is assembled in the chloroplast from eight molecules of 5-aminolevulinic acid, a highly reactive nonprotein amino acid (5-amino, 4-keto pentanoic acid). The bonds highlighted in red in the chlorophyll structure of Figure 1 delineate the locations of the atoms derived from the eight 5-aminolevulinic acid carbon skeletons. An intact 5-aminolevulinate molecule can be recognized in pyrrole ring IV: the nitrogen coordinated with the magnesium atom originates from the amino group; it is followed by four carbon atoms and the carboxyl-group, which is esterified to phytol.

Biosynthesis of chlorophyll comprises a number of challenging topics. Because mutants play an increasing role in exploring the biochemistry and molecular biology of this pathway and its regulation, we first highlight recent results on the use of mutants. The subsequent sections deal with the biosynthesis of 5-aminolevulinate, the synthesis of the pyrrole ring, the synthesis of the tetrapyrrole chain, and the formation of the tetrapyrrole ring. We then discuss trimming of the acetate and propionate side chains and establishment of the conjugated double bond system; insertion of the magnesium, formation of ring V, and synthesis of protochlorophyllide; and reduction to chlorophyllide and phytylation. Next, we discuss chlorophyll synthesis in chloroplast development and end with a discussion of chlorophyll binding proteins.

EXPLORING CHLOROPHYLL SYNTHESIS WITH MUTANTS

When barley grains are germinated in the dark, the seedlings have yellow leaves because they lack chlorophyll and contain only small amounts of protochlorophyllide (Figure 2A). If dark-germinated seedlings are supplied with 5-aminolevulinate in the dark, they will green in the course of a few hours due to the accumulation of protochlorophyllide, the next-to-last precursor for chlorophyll that in barley and other angiosperms requires light and binding to protochlorophyllide reductase to be converted into chlorophyllide, which is then phytylated to chlorophyll a (see later discussion). The accumulation of large amounts of protochlorophyllide reveals that all enzymes required to convert 5-aminolevulinate into chlorophyll are present in the plastids of dark-grown leaves. The formation of 5-aminolevulinate thus is limiting in the etioplasts. Mutations in four barley genes (\( tigrina-b, -d, -n, \) and -o) remove this limitation and accumulate two to 10 times the wild-type amount of protochlorophyllide in the dark (Nielsen, 1974). The homozygous mutant seedlings develop a 10-cm-high primary leaf but die after 10 to 15 days, when the nutrient supply from the grain endosperm is exhausted. One of these mutants, \( tigrina-d^{12} \) (Figure 2B), has no other defect than unregulated 5-aminolevulinate formation and therefore excessive protochlorophyllide synthesis in the dark. Homozygous \( tigrina-d^{12} \) mutants are fully viable and fertile if grown in continuous light but are dark sensitive. The dark sensitivity accounts for the name of these mutants: in light/dark cycles, the seedlings are green-white banded (Figures 2D, \( tigrina-b^{19} \) and 2E, \( tigrina-d^{12} \)). The white, often necrotic leaf domains are caused by the accumulation of protochlorophyllide in the dark. Only a normal amount of protochlorophyllide reductase is available to convert the protochlorophyllide into chlorophyllide, and when the light turns on, the excess of protochlorophyllide causes photodynamic damage to the plastids. With short light pulses, the accumulated protochlorophyllide can be converted successively into chlorophyll, and the \( tigrina-d^{12} \) mutant can be saved. This is not so with other mutants, such as \( tigrina-b^{19} \), which accumulate \( \zeta \)-carotene in addition to protochlorophyllide (Nielsen and Gough, 1974). Mutations in the \( tigrina-n \) and -o genes deregulate the synthesis of \( \beta \)-carotene as well as protochlorophyllide, with the result that lycoponic pigments accumulate and chloroplasts form.

Another class of mutants with disrupted chlorophyll biosynthesis is the yellow \( xantha \) mutants of barley. Using
complementation tests, five xantha genes have been identified that upon mutation block the pathway between 5-aminolevulinate and protochlorophyllide (see later discussion). When dark-grown leaves of the xantha-P mutant are fed 5-aminolevulinate, they accumulate a brown-red pigment (Figure 2C) that is a mixture of Mg-protoporphyrin and its monomethyl ester, just like xantha-135 seedlings fed 5-aminolevulinate. Therefore, we accumulate Mg-protoporphyrin and its monomethyl ester, just as Mg-protoporphyrin monomethyl ester. Using probes from the Rhodobacter gene, efforts are currently under way to clone the corresponding barley gene, which may correspond to the xantha-I or viridis-k gene.

Mutants at the loci xantha-I (Figure 2D), xantha-g, and xantha-h (Figure 2E) accumulate protoporphyrin IX when fed 5-aminolevulinate (von Wettstein et al., 1971, 1974). The products of these three genes are thus required for the insertion of Mg2+ into protoporphyrin IX. Disruption of any one of the three open reading frames bchD, bchl, and bchH of Rhodobacter capsulatus by interposon mutagenesis also leads to the secretion of protoporphyrin IX (Bollivar et al., 1994b), which suggests that their products are necessary for the Mg-chelatase reaction. When the homologous bchD, bchl, and bchH genes from R. sphaeroides are expressed separately in Escherichia coli, three proteins of 70, 40, and 140 kD, respectively, are produced. The BchD protein aggregates to form a 650-kD homopolymer. When combined in vitro, the 650-, 40-, and 140-kD proteins catalyze the insertion of Mg2+ into protoporphyrin IX in an efficient, ATP-dependent manner, thus confirming their biological function (Gibson et al., 1995).

Nuclear genes corresponding to bchl and bchH have now been identified in higher plants (Koncz et al., 1990; Hudson et al., 1993; P.F. Jensen, R.D. Willows, B. Larsen-Petersen, C.G. Kannangara, B.M. Stummann, D. von Wettstein, and K.W. Henningsen, manuscript submitted). Thus, four barley xantha-h mutants lack the Bchl-immunoreactive protein, and cloning of the xantha-h gene showed it to encode a protein with ~50% amino acid sequence identity to the Rhodobacter 40-kD bchl subunit and 85% to the deduced protein sequence of the Arabidopsis chlororap (ch-42) gene. The Arabidopsis and barley genes encoding this Mg-chelatase subunit are located in the nuclear genome, whereas in Euglena the gene is found in the chloroplast genome (Orsat et al., 1992). From an evolutionary point of view, it is interesting that the primary structures of the nonidentical domains of this protein are more closely conserved between the higher plant and Euglena subunits than between the Euglena and Rhodobacter subunits.

In two xantha-f mutants, no protein was detectable with antibodies directed against the higher plant homolog of the 140-kD Rhodobacter BchH protein. By exploiting nucleotide sequence homology to the bchH gene from Rhodobacter, the barley gene was cloned. It contains three introns and codes for a 1381-amino acid–long protein (153 kD), the N-terminal 50 residues of which are considered to target the precursor of the mature subunit from its site of synthesis in the cytosol into the chloroplast. In these two xantha-f mutants, no significant amount of transcripts from this gene were found (P.F. Jensen, R.D. Willows, B. Larsen-Petersen, C.G. Kannangara, B.M. Stummann, D. von Wettstein, and K.W. Henningsen, manuscript submitted), which indicates that the barley xantha-f gene is the structural gene for the 140-kD subunit of Mg-chelatase. The barley protein displays 82% amino acid sequence identity to the olive protein of snapdragon (Hudson et al., 1993), 66% identity to the cyano bacterial Synechocystis protein (Jensen et al., 1995), and 34% identity to the Rhodobacter BchH subunit. The snapdragon olive mutations were recognized as yellow-green leaf variegations, and the gene was subsequently cloned with the aid of a known transposon insertion that had inactivated the gene. The hunt is on for the gene encoding the third (70 kD) subunit of higher plant Mg-chelatase.

**Figure 1.** The Chemical Structure of Chlorophyll a.

Every chlorophyll molecule is synthesized in the chloroplast from eight molecules of 5-aminolevulinic acid. The eight red heavy lines indicate the location of the atoms derived from these molecules of 5-aminolevulinic acid in the finished molecule. Position 3, which is occupied by a methyl group in chlorophyll a and a formyl group in chlorophyll b, is indicated.
Figure 2. Barley Mutants in Structural and Regulatory Nuclear Genes Involved in the Synthesis of Chlorophyll.

(A) Primary leaves of barley grown for 7 days in darkness at 23°C.
(B) Primary leaves of the mutant tigrina-d112 grown for 7 days in darkness. The green pigmentation is due to unregulated accumulation of protochlorophyllide. A phenocopy of the mutant can be obtained by feeding wild-type leaves for 24 hr with 5-aminolevulinate.
(C) Primary leaves of the mutant xantha-f° fed for 24 hr with water (left) or 0.01 M 5-aminolevulinate (right). These mutants are unable to carry out the cyclase reaction to form ring V of the chlorophyll molecule and thus accumulate red Mg-protoporphyrin IX and its monomethyl ester.
(D) F2 generation segregating a homozygous xantha-f° seedling (yellow) and a homozygous tigrina-b9 mutant. The double mutant is distinguishable from xantha-f° by photometrically detectable overproduction of protoporphyrin IX.
(E) F2 generation segregating homozygous xantha-hs7 seedlings (yellow) unable to insert Mg2+ into protoporphyrin IX and homozygous tigrina-d12 seedlings (tiger banded). Double mutants are yellow but do not accumulate protoporphyrin IX.

The possibility to express active Mg-chelatase subunits from bacteria and higher plants in heterologous hosts in large quantities makes it possible to study how insertion of Mg2+ into the protoporphyrin is carried out by the three subunits, a problem that could not be addressed previously. The mechanism appears to be related to the insertion of other ions into the protoporphyrin ring: cobaltochelatase of *Pseudomonas denitrificans*, which inserts Co to yield cobyricin acid-a, c-diamide in vitamin B12 synthesis, also consists of three polypeptides (Debussche et al., 1992). The porphyrin binding 140-kD cobN
The gene product shows homology to the large subunit of Mg-chelatase, whereas the other two required polypeptides, CobS and CobT (38 and 80 kD), have a similar size but no sequence homology to Bchl and BchD. We surmise that the large subunit in Mg-chelatase also will turn out to carry the porphyrin binding pocket.

In cases in which mutants in steps of chlorophyll synthesis are not obtainable or viable at the seedling stage, information about the function of genes cloned by oligonucleotides derived from amino acid sequences of purified enzymes of the pathway from any organism can be obtained by overexpressing them or by expressing antisense genes using either the cauliflower mosaic virus 35S promoter or a tissue-specific promoter. Figure 3 provides an example of this approach, in which tobacco plants were transformed with an antisense glutamate 1-semialdehyde aminotransferase gene. This enzyme catalyzes the last step in 5-aminolevulinate synthesis (see subsequent discussion), and antisense transformants show a general or tissue-specific reduction in chlorophyll (Höfgen et al., 1994). Partial or complete suppression of the aminotransferase mimics a wide variety of inheritable chlorophyll variegation patterns elicited by nuclear and organelle gene mutations in different higher plants. Such antisense plants provide a sensitive visible marker for activities of chlorophyll synthesis genes in leaves, stems, and flowers. The cell- or tissue-specific action of such antisense genes depends inter alia on the position of insertion in the chromosomes of the host, a position we at present cannot control. However, transgenic plants blocked in specific steps in chlorophyll synthesis provide the experimental material to address important questions. For example, translation of chlorophyll binding proteins on chloroplast ribosomes is halted unless chlorophyll is present and can bind to the nascent polypeptide chains. If chlorophyll synthesis is limited by the expression of an antisense gene, does translation of chlorophyll binding proteins on chloroplast ribosomes then take preference over translation of chlorophyll binding proteins on cytosolic ribosomes and their import into the organelle? We would also like to know if angiosperm cells can synthesize haem with a 5-aminolevulinate synthase when they are prevented from producing 5-aminolevulinate from glutamate by complete antisense inhibition of glutamate semialdehyde aminotransferase.

Antisense or sense approaches have led to transgenic tobacco plants with reduced glutamate-tRNA synthase (Andersen, 1992) and reduced glutamyl-tRNA reductase (B. Grimm, personal communication). Like the plants with reduced glutamate 1-semialdehyde aminotransferase (Figure 3), the transgenic plants exhibited reduced chlorophyll content and variegation. Tobacco antisense plants with reduced uroporphyrinogen decarboxylase (Mock et al., 1995) and reduced coproporphyrinogen III oxidase display extensive necrotic lesions on the leaves, presumably because they accumulate photosensitive porphyrin precursors (Kruse et al., 1995a, 1995b).

SURPRISES IN THE BIOSYNTHESIS OF 5-AMINOLEVULINATE

There are two distinct routes for the synthesis of 5-aminolevulinate, one utilizing a condensation reaction of glycine with succinyl-CoA by the enzyme 5-aminolevulinic acid synthase.

Figure 3. Tobacco Transformants Expressing a Glutamate 1-Semialdehyde Aminotransferase Antisense Gene.

The transformant at left has pale leaves, whereas in the transformant at center, chlorophyll formation is inhibited along the leaf veins. The plant at right is the untransformed control. From Höfgen et al. (1994) and used with permission of the National Academy of Sciences.
and the other a three-step pathway from glutamate that is called the C₅ pathway (Jordan, 1991). The first route is used by animals, yeast, and a number of bacteria, notably *Rhodobacter*, *Rhodospirillum*, and *Rhizobium*, whereas the C₅ pathway is characteristic of higher plants, bryophytes, cyanobacteria, and many eubacteria. In the phytophagellate *Euglena gracilis*, the two pathways are used in different compartments. The C₅ pathway operates in chloroplasts and is exclusively responsible for chlorophyll synthesis there, whereas in mitochondria, 5-aminolevulinate synthase is responsible for the synthesis of heme a of cytochrome c oxidase (Weinstein and Beale, 1983).

When greening plants or algae are treated with levulinate, 5-aminolevulinic acid accumulates because levulinate inhibits the enzyme 5-aminolevulinate dehydratase (Beale and Castelfranco, 1974a). When 14C-labeled glutamate is fed together with levulinate, the radioactivity is found in the accumulated 5-aminolevulinic acid (Beale and Castelfranco, 1974b). By using glutamic acid labeled with 14C in different positions as precursors and determining the label distribution in the resulting 5-aminolevulinic acid or in the entire chlorophyll molecule, it was shown that the intact five-carbon skeleton of glutamate is incorporated into 5-aminolevulinic acid (Beale et al., 1975; Meller et al., 1975; Porra, 1986).

Figure 4. Biosynthesis of 5-Aminolevulinic acid.

It was a great surprise when it was discovered that conversion of glutamate to 5-aminolevulinate requires glutamate to be activated at the α-carboxyl by ligation to tRNA<Glu>. It was equally surprising that the *hema* gene of *E. coli* (Sasarman et al., 1968) encodes not 5-aminolevulinic acid synthase, as had been thought for 20 years, but glutamyl-tRNA dehydrogenase. Moreover, a second *E. coli* gene, identified as giving rise upon mutation to 5-aminolevulinic acid auxotrophy (Powell et al., 1973), turned out to be a remarkable enzyme: glutamate-semialdehyde aminotransferase.

As detailed in Figure 4, glutamic acid is first activated by ligation to tRNA<Glu> with an aminocarboxyl-tRNA synthetase in the presence of ATP and Mg²⁺. The activated glutamate is then reduced to glutamate 1-semialdehyde in an NADPH-dependent reaction catalyzed by Glu-tRNA reductase. Glutamate 1-semialdehyde-2,1-aminomutase (EC 5.4.3.8) then carries out the conversion into 5-aminolevulinate. Because we have recently provided a detailed account of our present knowledge of this pathway with relevant references (Kannangara et al., 1994), in this article we highlight additional interesting elements and novel observations of importance for further work.

In higher plants, the gene encoding the tRNA<Glu> is encoded in chloroplast DNA, whereas the three enzymes involved in 5-aminolevulinic acid formation—the aminocarboxyl-tRNA synthetase, the reductase, and the aminotransferase—are encoded by nuclear DNA and are imported into the chloroplast stroma after synthesis by cytoplasmic ribosomes. This single chloroplast tRNA<Glu> has to serve for both chlorophyll synthesis and protein synthesis on chloroplast ribosomes. In the presence of ATP and glutamate, purified Glu-tRNA synthetase, tRNA<Glu>, and Glu–tRNA reductase can form a complex; this implies that different domains of the tRNA<Glu> molecule recognize and bind to the two enzymes. This conclusion has recently been verified by studies of a *E. gracilis* mutant in which a cytosine of the T-loop of tRNA<Glu> was converted to a uracil (Stange-Thomann et al., 1994). This tRNA can still be charged with glutamate by the aminocarboxyl-tRNA synthetase, as judged by the capacity of the mutant to synthesize ribulose-bisphosphate carboxylase and ATP synthase on chloroplast ribosomes, but it is unable to function with the reductase in the synthesis of 5-aminolevulinic acid.

The ligation and reduction reactions require both the 3'CCA sequence and the UUC anticodon with its hypermodified 5-methylaminomethyl-2-thiouridine of tRNA<Glu>. Although the ligase does not discriminate between tRNA<Glu> and tRNA<Asn> and loads glutamate onto both tRNA species, the reductase recognizes glutamyl-tRNA<Asn> exclusively (glutamyl-tRNA<Asn> is converted to glutamyl-tRNA<Asn> by a specific aminotransferase). Chloroplast glutamyl-tRNA<Glu> from barley,
wheat, tobacco, spinach, cucumber, Synechocystis, and Chlamydomonas are equally efficient as substrates for the barley glutamyl-tRNA reductase, but *E. coli* tRNA\textsubscript{Glu} is a much poorer substrate, and yeast tRNA\textsubscript{Glu} is only slightly better (Willows et al., 1995). A comparison of the bases conserved among tRNA\textsubscript{Glu} and tRNA\textsubscript{Gln} from different species and cell compartments reveals that five nucleotides are probably required for recognition by the glutamyl-tRNA synthetase. An analogous comparison of sequences that can and cannot be used by the reductase indicates that seven nucleotides are involved in recognition by the barley chloroplast glutamyl-tRNA\textsubscript{Glu} reductase.

Two approaches have been taken to characterize the latter unique enzyme, which converts glutamyl-tRNA\textsubscript{Glu} to glutamate 1-semialdehyde (reviewed in Pontoppidan and Kannangara, 1994). One approach was to purify this unstable enzyme of low abundance, whereas the other centered around mutations in the *hemA* gene of the *E. coli* K12 strain which leads to auxotrophy for 5-aminolevulinate. Recently, unequivocal evidence was obtained that *hemA* encodes the glutamyl-tRNA\textsubscript{Glu} reductase and that no other protein is required for activity (Pontoppidan and Kannangara, 1994). The purified barley enzyme, with a molecular mass of \( \approx 270 \) kD, is made up of four to six identical subunits with a molecular weight of \( 54 \) kD and has an activity for synthesis of glutamate semialdehyde of 250 pmol min\(^{-1}\)\mu g\(^{-1}\). The sequence of the 18 N-terminal amino acids of the mature subunits of the active enzyme is identical to the N-terminal amino acid sequence deduced from a barley cDNA clone that rescues the *E. coli* hemA mutant. Complementation of the *E. coli* mutant has yielded over the years *hemA* genes from *Salmonella typhimurium*, *Bacillus subtilis*, *Synechocystis*, Arabidopsis, *Clostridium josni*, barley, and other species. Uncertainty has reigned about the function of the *HemA* gene product for two reasons. (1) The mutant can also be rescued by the genes encoding the monomeric 5-aminolevulinate synthase from *Rhodobacter* and *Rhizobium*, the enzyme using the ubiquitous metabolites succinyl-CoA and glycine as substrates. When the *E. coli* *hemA* gene was cloned and sequenced, its deduced amino acid sequence showed no homology to that of 5-aminolevulinate synthase from *Rhodobacter* and mouse. At the same time, identification of the *E. coli* hemL gene as the structural gene for glutamate 1-semialdehyde aminotransferase and demonstration that this enzyme as well as glutamyl-tRNA synthetase are active in the *E. coli* *hemA* mutant established that *E. coli* uses the C\(_5\) pathway. This is a good illustration that complementation of a mutant with expression of a known enzyme does not necessarily identify the enzymatic function of the cognate wild-type gene. (2) Although *hemA* genes from several bacteria and higher plants can complement the *E. coli* hemA mutant and cause *E. coli* cells to overproduce uroporphyrinogen, HemA protein purified from *E. coli* overexpressing the *hemA* gene is not enzymatically active in vitro.

The deduced amino acid sequences of eight glutamyl-tRNA\textsubscript{Glu} reductases from a range of organisms contain several highly conserved sequence domains, including one of 27 residues that is 70% identical in all eight proteins. Analysis of the function of this domain and elucidation of the mechanism of the reduction of glutamyl-tRNA\textsubscript{Glu} requires establishing an expression system yielding high amounts of active enzyme.

Barley contains two *hemA* genes, which have different expression patterns. One gene is not transcribed in roots but gives a high transient transcript level during greening; a second is expressed moderately in roots and leaves (O. Bougri and B. Grimm, personal communication). Light-stimulated transcription of the Arabidopsis genes for glutamyl-tRNA\textsubscript{Glu} reductase and glutamate semialdehyde aminotransferase has been reported by Idag et al. (1994). Further experiments are necessary to determine to what extent the regulation of 5-aminolevulinate synthesis takes place at the transcriptional and/or translational level.

Glutamate 1-semialdehyde aminotransferase transfers the amino group from the C4 carbon of glutamate semialdehyde to the C5 carbon of 5-aminolevulinic acid, with pyridoxal 5' phosphate as a cofactor and 4,5-diaminovalerate as an intermediate (Kannangara et al., 1994). Overexpression and purification of recombinant enzyme from *Synechococcus* and barley and site-directed mutagenesis have permitted a detailed investigation of the ping-pong bi-bi mechanism by which the transamination is effected. The 46-kD enzyme of *Synechococcus* can function as a monomer, whereas the barley enzyme operates as homodimeric protein. X-ray crystallography is presently being employed to study the interaction between the two subunits (Hennig et al., 1994).

### THREE REMARKABLE ENZYMES GENERATE UROPORPHYRINOGEN III

The biosynthetic pathway of chlorophyll from 5-aminolevulinate to uroporphyrinogen III and beyond to protoporphyrin IX is, according to our present knowledge, essentially the same as that of heme in man, mouse, plants, and bacteria. Much more molecular information for plants is desirable, and the present discussion endeavors to emphasize molecular results in mammals and bacteria that might be confirmed for plants.

The carbon-carbon and carbon-nitrogen bonds in the pyrrole ring of porphobilinogen are formed by the enzyme 5-aminolevulinic acid dehydratase (porphobilinogen synthase), which catalyzes the asymmetric condensation of two molecules of 5-aminolevulinic acid, as shown in Figure 5 (Jordan, 1991; Spencer and Jordan, 1994). Isotopically labeled substrates reveal that the initially bound 5-aminolevulinic acid forms a Schiff base with its keto group and the \( \epsilon \)-amino group of a lysine at position 247 of the enzyme (*E. coli* numbering) that, along with an adjacent proline, is conserved in all 16 dehydratases whose sequence is known. These enzymes include representatives from mammals, bacteria, yeast, and plants, including spinach (Schaumburg et al., 1992), pea (Boese et al., 1991), tomato, and soybean. This first bound molecule gives rise to the propionic acid side chain of
Chlorophyll Biosynthesis

Figure 5. Synthesis of the Pyrrole Ring.

Figure 6. Synthesis of the Tetrapyrrole Chain.
The dipyrromethane cofactor is located in a cleft between domain 1 (blue) and domain 2 (green) and is covalently bound to Cys-242 of domain 3 (violet). From Lambert et al. (1994).

turned around (Figure 8; Battersby et al., 1977). This can be seen in the structure of uroporphyrinogen III and the finished chlorophyll molecule because the methyl groups of ring IV and I are located in neighboring positions within the porphyrin ring. The enzymes of human, mouse, E. coli, B. subtilis (Hansson et al., 1991), and Synechococcus (Jones et al., 1994) have a molecular weight of ~30 kD and are highly diverse in primary structure. Only five residues are conserved among the nine enzymes for which the sequence is known.

**TRIMMING OF THE SIDE CHAINS AND RED COLOR FORMATION**

Figure 9 presents the following steps of chlorophyll formation: the acetic acid side chains are shortened to methyl groups by uroporphyrinogen III decarboxylase, coproporphyrinogen III oxidase trims two of the propionic acid side chains into vinyl groups, and then the molecule is oxidized by protoporphyrinogen IX oxidase to establish the conjugated double-bond system of the red-colored protoporphyrin IX (cf. Akhtar, 1994).

Uroporphyrinogen III decarboxylases from mammalian erythrocytes, and liver, yeast, Rhodobacter sphaeroides (Jones and Jordan, 1993), B. subtilis (Hansson and Hederstedt, 1992), and the stroma of tobacco and barley plastids (Mock et al., 1995) are monomeric proteins with a molecular weight of ~40 kD. At low-substrate concentrations, the enzyme catalyzes decarboxylations, beginning at ring IV, proceeding clockwise through rings I and II, and ending at ring III. At high-substrate concentrations, decarboxylation can occur in any order. The 10 primary amino acid sequences show that the eukaryotic enzymes have an overall similarity of 33 to 50%. The tobacco and barley sequences are 73% similar to the sequence of the cyanobacterial (Synechococcus) enzyme (Kiel et al., 1992). The sequence comparisons indicate that there are three invariant domains. Their functions, and those of individual invariant amino acids, remain to be determined.

Structural genes for oxygen-dependent coproporphyrinogen III oxidases from three higher plants, including soybean (Madsen et al., 1993), barley, and tobacco (Kruse et al., 1995), have been cloned. Coproporphyrinogen III oxidase is synthesized as a precursor protein on cytoplasmic ribosomes and is imported into the stroma of pea chloroplasts, where it is processed into a catalytically active enzyme with a molecular weight of 39 kD. The three higher plant enzymes are 70% identical at the amino acid sequence level, and an overall comparison from bacteria to human of the eight known primary structures shows 50% identity, which is especially prominent in the C-terminal part of the oxidase. Levels of transcripts for the plant coproporphyrinogen III oxidase are the same in dark-grown and greening leaves of barley; they are also constant during leaf development in tobacco but are strongly reduced in mature leaves. In anaerobic bacteria synthesizing bacteriochlorophyll, the oxygen-requiring coproporphyrinogen III oxidase must be substituted by an oxygen-independent enzyme. A mutant in R. sphaeroides excreting coproporphyrin III has defined a gene encoding a putative anaerobic coproporphyrinogen oxidase (Coomber et al., 1992) that is also present in E. coli, Salmonella, Pseudomonas, Rhizobium, and B. subtilis (cf. Hansson and Hederstedt, 1994).

Protoporphyrinogen IX oxidase has been purified to homogeneity from bovine and mouse liver (cf. Akhtar, 1994) and barley etioplasts (Jacobs and Jacobs, 1987). The mammalian enzymes require molecular oxygen, use a flavin cofactor, and have a molecular mass of 65 kD, whereas the barley enzyme is 36 kD. Expression of the hemY gene of the B. subtilis hemEHY cluster in E. coli results in the production of a soluble 53-kD protein that can oxidize both coproporphyrinogen III and protoporphyrinogen IX (Dailey et al., 1994; Hansson and Hederstedt, 1994). Cloning of the gene for the chloroplast
protoporphyrinogen IX oxidase and its heterologous expression will help verify the catalytic mechanism, which consists of three consecutive dehydrogenations and a subsequent tautomerization yielding the porphyrin through the stereospecific loss of the fourth meso hydrogen as a proton (Akhtar, 1994). In the three desaturations, the flavin prosthetic group acts as an acceptor of the hydride ion; the reduced flavin is then reoxidized by \( \text{O}_2 \) for regeneration of active enzyme.

FROM BRIGHT RED TO GREEN

The chlorophyll- and bacteriochlorophyll-specific pathway from protoporphyrin IX to chlorophyll (Figure 10) begins with the insertion of the Mg\(^{2+}\) ion (Bauer et al., 1993; Bollivar et al., 1994b; Castelfranco et al., 1994). Recent advances in the characterization of this reaction were described earlier, in the discussion of the use of mutants. After insertion of the Mg\(^{2+}\) ion, the magnesium–protoporphyrin methyltransferase esterifies the propionic side chain of ring III in preparation for the cyclization reaction that produces ring V. The S-adenosyl-L-methionine that donates the methyl group must be imported into the chloroplast, which also contains a methyl esterase and an oxidase to degrade unused Mg-protoporphyrin IX monomethyl ester (Castelfranco et al., 1994). Expression of the bchM gene of \( R. \) capsulatus and \( R. \) sphaeroides in \( E. \) coli have identified this gene’s product as the methyltransferase for Mg-protoporphyrin IX (Bollivar et al., 1994a; Gibson and Hunter, 1994). \(^{18}\text{O}_2\) labeling indicates that the oxygen atom in ring V derives from atmospheric \( \text{O}_2 \) and that \( \beta \)-hydroxypropionate

![Figure 9. Trimming of the Acetate and Propionate Side Chains and Establishment of the Conjugated Double Bond System.](image-url)

![Figure 10. Pathway from Protoporphyrin IX to Chlorophyll a.](image-url)
and β-oxopropionate are intermediates in the cyclization process (Castelfranco et al., 1994).

Reduction of divinyl protochlorophyllide to monovinylprotochlorophyllide has been inferred from product characterization (Whyte and Griffiths, 1993). Because disruption of the bchJ gene in R. capsulafus leads to accumulation of divinyl protochlorophyllide, this gene is thus a candidate for a structural gene for this enzyme (Bollivar et al., 1994b).

Protochlorophyllide reduction in plants and bacteria has been studied extensively and is the topic of the next section. Briefly, the transfer of a hydrogen from NADPH to the carbon atom carrying the propionic side chain of ring IV and a hydrogen from water or protein to the carbon atom carrying the methyl group by protochlorophyllide reductase is carried out in angiosperms by a photochemical reaction. In cotyledons of gymnosperms and in mosses, liverworts, algae, and photosynthetic bacteria, the reduction can be catalyzed by a very different enzyme in the dark.

The last step in the synthesis of chlorophyll a is catalyzed by chlorophyll synthetase. This enzyme esterifies the propionic acid side chain of ring IV with either phytyl pyrophosphate, as is preferred in chloroplasts, or geranylgeranyl pyrophosphate, with subsequent reduction of the double bonds at positions 6, 10, and 14, as is preferred in etioplasts (Rüdiger et al., 1980). Nuclear magnetic resonance analysis of bacteriochlorophyll synthesized by R. sphaeroides from 5-amino-levulinic acid labeled at the carboxyl group with 13C and 18O revealed retention of both 18Os in the formation of the ester bond and thus a carboxyl–alkyl transfer mode (cf. Akhtar, 1994). In Rhodobacter, the products of two genes (bchG and bchP) are implicated in the phytolation of bacteriochlorophyll a.

After many years of debate, there is now strong evidence that the formyl group in chlorophyll b that replaces the methyl group in position 3 (cf. Figure 1) of chlorophyll a is inserted by an oxygenase enzyme. Mass spectrometry of chlorophyll b synthesized in dark-grown maize leaves in the presence of 13C and 18O has documented that the oxygen in the formyl group of chlorophyll b is incorporated exclusively from atmospheric O2 (Porra et al., 1994), as is the case in Chlorella (Schneegurt and Beale, 1992). Oxygenation of chlorophyll a by a single oxygenase is supported by the observation that 10 independently isolated chlorophyll b-deficient mutants in barley and 54 b-less mutants in Chlamydomonas belong to a single complementation group (Simpson et al., 1985; Chunayev et al., 1991).

BACTERIA-INVENTED PROTOCHLOROPHYLLIDE REDUCTION WITH AND WITHOUT LIGHT

Rhodobacter reduces protochlorophyllide to chlorophyllide in the dark, using an enzyme consisting of three polypeptides, encoded by bchL, bchN, and bchB (Burke et al., 1993a). Elimination of the double bond in ring IV in the course of protochlorophyllide reduction is similar to elimination of the double bond in ring II by chlorin reductase, which converts chlorophyllide a into 2-desacyetyl-2 vinyl bacteriochlorophyllide a. Chlorin reductase comprises the products of the bchX, bchY, and bchZ genes. BchX and BchL share 34% amino acid sequence identity with each other, as well as 30 to 37% identity with the Fe protein (NifH) of the Acetobacter vinelandii nitorgenase (Burke et al., 1993b). The nucleotide binding site and the 4Fe-4S cysteine binding ligands are especially conserved in all known BchL subunits (Koonin, 1993). In addition, bchN is homologous to nifK, the gene encoding the β-subunit of the MoFe protein of nitorgenase, with its 4Fe-4S cluster pair. The three cysteine ligands of NifK are conserved in the BchN sequences. The bchB product is homologous to the Vd-Fe alternative α subunit of nitorgenase (nifD). However, the FeMo binding ligands Cys-275 and His-442 are replaced by tyrosines, indicating that another cofactor reduces protochlorophyllide.

Genes encoding the three polypeptides that are implicated in light-independent protochlorophyllide reduction have been identified in the cyanobacteria Plectonema boryanum, Synchocystis, and Synechococcus (Li et al., 1993; Suzuki and Bauer, 1995). Genes encoding these polypeptides have also been identified in the chloroplast genomes of the liverwrt Marchantia polymorpha, Chlamydomonas reinhardtii, C. moewusii, the red alga Porphyra purpurea, ferns, and gymnosperms (Li et al., 1993; Suzuki and Bauer, 1995), all of which are known to synthesize chlorophyll in the dark. Disruption of the Chlamydomonas bchL homolog by particle gun transformation produces a "yellow in the dark" phenotype; that is, protochlorophyllide reduction in the dark is blocked (Suzuki and Bauer, 1992). The presence of these genes has been documented in the chloroplast DNA of the ferns Cystopteris fragilis and Athyrium filix femina; the horsetail Equisetum arvense; and the coniferous trees Ginkgo biloba (Richard et al., 1994), Pseudotsuga menziesii, Taxus, Juniperus, Araucaria, Pinus cortorta, and Picea abies. The genes are, however, absent in the chloroplast genome of the angiosperms tobacco, rice, maize, Arabidopsis, and Bougainvillia glabra, all of which are incapable of synthesizing chlorophyll in the dark.

In contrast to lower plants and gymnosperms, angiosperms use exclusively the nuclear-encoded light- and NADPH-dependent protochlorophyllide oxidoreductase to synthesize chlorophyllide. The barley and Arabidopsis enzymes have a molecular weight of 35 kD and are synthesized in the cytosol with transit peptides for import into etioplasts and chloroplasts. After expression of the plant cDNA genes in E. coli, the extracted enzyme was able to reduce the substrate protochlorophyllide only in the presence of light and NADPH (Schulz et al., 1989; Benli et al., 1991). Monovinyl and divinyl protochlorophyllide are equally acceptable as substrates (Knaust et al., 1993). Light-dependent protochlorophyllide reductases from barley, oats, wheat, pea, and Arabidopsis are 80 to 95% identical at the amino acid sequence level (Suzuki and Bauer, 1995) but bear no homology to the three peptides of the light-independent protochlorophyllide reductase.

Barley contains two genes encoding light-dependent protochlorophyllide oxidoreductase isoenzymes that are 75% identical.
at the amino acid sequence level (Holtorf et al., 1995). One
isoenzyme is abundantly synthesized in the dark, but its
synthesis declines rapidly upon illumination and greening of
dark-grown seedlings. Transcription of the gene for this iso-
enzyme is turned off upon illumination. The gene for the other
isoenzyme is constitutively transcribed in the dark and in the
light, and the transcripts are translated continuously into ac-
tive enzyme, which is responsible for chlorophyll synthesis
during greening. This explains why transcripts for the light-
dependent protoclorophyllide reductase as well as the pro-
tein itself were found to be abundant in dark-grown seedlings
of bean, pea, tomato, sunflower, mustard, Arabidopsis, and
maize, but both mRNA and enzyme levels declined when the
seedlings were illuminated and rapid chlorophyll synthesis
ensued (Forreiter et al., 1990; Benli et al., 1991). These experi-
ments, as it turns out, focused on the “wrong” isoenzyme.

In gymnosperms such as pine and spruce, the chloroplast
genome encodes the light-independent protoclorophyllide
reductase and the nuclear genome two light-dependent pro-
toclorophyllide reductases. The light-independent enzyme is
expressed only during development of the cotyledones in the
dark but not during development of the primary and second-
ary needles, in which chlorophyll synthesis relies on the
light-dependent protoclorophyllide reductase. As in barley,
there are two light-dependent isoenzymes in Pinus mugo,
P. strobus, and P. taeda; one is synthesized specifically in the
dark and the other constitutively in the dark as well as during
chloroplast development in the light (Forreiter and Apel, 1993).
Both of them are synthesized in the cotyledons together with
the light-independent enzyme.

It was originally thought that the light-dependent proto-
clorophyllide oxidoreductase was an evolutionary invention
of angiosperms. However, in a seminal experiment that illus-
trates the power of using Rhodobacter mutants in the analysis
of chlorophyll biosynthesis genes and enzymes from cyanobac-
teria and higher plants, Suzuki and Bauer (1995) demonstrate
that cyanobacteria had probably already invented the light-
dependent enzyme before the advent of angiosperms. An
R. capsulatus mutant that is unable to reduce protoclorophyl-
lide as a consequence of the disruption of the bchL gene was
mated with an E. coli strain library containing cosmids of 19
gene equivalents of Synechocystis (strain PCC6803) under
a strong Rhodobacter promoter. Some exconjugants selected
under anaerobic light conditions permitted the synthesis of
bacteriochlorophyll and photosynthesis. The Synechocystis
bchL gene that conferred the ability for light-dependent protochloro-
pseudoylide:NADPH-oxidoreductase complex (Henningsen et al., 1993).

The protochlorophyllide in the prolamellar bodies is reduced
to chlorophyllide, and the membrane tubes lose their ordered
arrangement (Figure 11B) and flow out into fenestrated primary
lamellar layers (Figure 11C). The dispersal of the prolamellar
body is preceded or accompanied within minutes by a spec-
tral shift of protoclorophyllide absorption from 684 to 672 nm
and esterification of chlorophyllide to chlorophyll a (Henningsen
et al., 1993). Chlorophyll synthetase seems to be present in
the cristalline prolamellar bodies but does not become ac-
active, at least in wheat, until the protoclorophyllide has been
reduced by light and the tubes have lost their regular configu-
ration (Lindsten et al., 1993). Over the next 24 hr, chlorophyll
is synthesized and the complexes of photosystem I and II and
cytochrome b6/f (Figure 12) are assembled. Incipient grana
are formed as overlapping, paired primary lamellar layers, and
respectable grana are present after 24 hr (Figure 11D).

Assembly of the crystalline configuration of the prolamel-
lar body requires the presence of protoclorophyllide and
NADPH. The xanthaFmutant, which is unable to synthesize
Mg-chlorophyllide, and other mutants stringently blocked in
protoclorophyllide synthesis contain only unfenestrated thyla-
koids (Figure 11F) and lack prolamellar bodies, even though
the 36-kD protoclorophyllide reductase protein is still synthe-
sized (Dehesh et al., 1986). Arabidopsis mutants with lesions
in the nuclear transcription factor DET1 also form expanded
leaves in the dark and etioplasts lacking prolamellar bodies.

**CHLOROPHYLL SYNTHESIS AND CHLOROPLAST
DEVELOPMENT**

Chloroplasts of higher plants develop from proplastids that
consist of envelope and internal vesicular membranes in ad-
dition to invaginations from the inner envelope membrane (von
Wettstein, 1958; Henningsen et al., 1993). In light-exposed tis-
sue, the proplastids grow in size and differentiate a mem-
brane system composed of unpaired stroma thylakoids (discike
sacs) that are continuous with paired or appressed thylakoids
in the grana (Figures 11D and 11E). The membranes of both types
of thylakoid contain chlorophyll bound to individual proteins.
If seedlings, especially those of the family Poaceae (for exam-
ple, barley, maize, and wheat), are germinated in the dark, the
plasts develop into etioplasts. These contain perforated pri-
mary lamellar layers and develop crystalline prolamellar bodies
(Figure 11A). The major protein constituent of the membranes
in the prolamellar body is a ternary protochlorophyllide:NADPH-
oxidoreductase complex (Henningsen et al., 1993). Upon illumi-
nation, the etioplasts synthesize chlorophyll, develop a lamellar
system, and become photoautotrophically active chloroplasts.

The protochlorophyllide in the prolamellar bodies is reduced
to chlorophyllide, and the membrane tubes lose their ordered
arrangement (Figure 11B) and flow out into fenestrated primary
lamellar layers (Figure 11C). The dispersal of the prolamellar
body is preceded or accompanied within minutes by a spec-
tral shift of protoclorophyllide absorption from 684 to 672 nm
and esterification of chlorophyllide to chlorophyll a (Henningsen
et al., 1993). Chlorophyll synthetase seems to be present in
the crystalline prolamellar bodies but does not become ac-
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reduced by light and the tubes have lost their regular configu-
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sized (Dehesh et al., 1986). Arabidopsis mutants with lesions
in the nuclear transcription factor DET1 also form expanded
leaves in the dark and etioplasts lacking prolamellar bodies.
Figure 11. Membrane Structures in Etioplasts, Developing Plastids, and Chloroplasts of Barley Seedlings.

(A) Etioplasts of 7-day-old dark-grown primary leaves contain crystalline prolamellar bodies. Protochlorophyllide oxidoreductase is the major protein in the prolamellar bodies.

(B) Upon transfer into light and reduction of protochlorophyllide to chlorophyllide, the membrane tubes of the prolamellar body lose their ordered arrangement and flow out into primary lamellar layers.
DET1 is required to initiate chloroplast development upon illumination (Chory et al., 1989; Pepper et al., 1994), and in det1 mutants, the synthesis of the protochlorophyllide reductase necessary for prolamellar body formation is probably prevented.

Reorganization and dispersal of the prolamellar body require protochlorophyllide and the capacity for phytylation. Thus, in senescing etioplasts, in which crystalline prolamellar bodies are depleted of protochlorophyllide, transformation into the unordered tubular configuration upon illumination is severely impeded. Several mutants in barley can be exploited to analyze the relationship between chlorophyll synthesis and development of membrane structures. In xantha-j mutants, protochlorophyllide oxidoreductase accumulates in normal amounts, crystalline prolamellar bodies are formed (Figure 11A), and reduction to chlorophyllide-684 and tube transformation are instantaneous upon illumination. However, dispersal of the prolamellar body, shift of the chlorophyllide absorption peak to 672 nm, and esterification by chlorophyll synthetase are blocked, and no further chloroplast development is possible (Henningsen et al., 1993). On the other hand, if the mutants are grown at low temperature in the light, ~10% of normal levels of chlorophyll and some grana can develop. Taking note of the recent discovery of two light-dependent protochlorophyllide reductases in barley, we suggest that the limited amount of chlorophyll formed in the light in the xantha-j mutants uses the constitutively expressed enzyme, whereas the dark-specific enzyme, with its bound photoreceptor protochlorophyllide, is located in the prolamellar bodies with the specific function to initiate the reorganization of this membrane storage body upon illumination. It is this reorganization that is defective in the xantha-j mutants.

Stacking of thylakoids into grana requires the presence of chlorophyll, and it has variously been suggested that thylakoid stacking is dependent on the presence of chlorophyll b, the light-harvesting complex LHCII, or the PSII complex (Simpson et al., 1989). However, even a double mutant in barley (chlorina-b, xantha-j) that lacks LHCII because of a block in the conversion of chlorophyll a to chlorophyll b and that is unable to assemble a functional inner core of photosystem II can still form grana (Simpson et al., 1989). The molecular basis for thylakoid stacking therefore remains unresolved.

When etiolated seedlings are returned to the dark after conversion of protochlorophyllide with a short illumination, protochlorophyllide is resynthesized and crystalline prolamellar bodies reassemble. Chloroplasts with both crystalline prolamellar bodies containing protochlorophyllide as well as many large, chlorophyll-containing grana develop in dark-grown barley seedlings illuminated for 24 hr with only 1 μE m⁻² sec⁻¹ of light. Upon illumination with higher intensity light, the protochlorophyllide is reduced and the prolamellar bodies disperse (Henningsen et al., 1993). The discovery of the two differentially regulated light-dependent protochlorophyllide reductases may explain the finding that the chlorophyll in the grana of the "low-light" chloroplasts is likely to be synthesized by the protochlorophyllide reductase isozyme that is constitutively expressed in the dark and during greening, whereas the "prolamellar body-specific reductase" maintains the prolamellar bodies under very low light intensity.

Crystaline prolamellar bodies and extensive grana are also found in chloroplasts of cotyledons of Norway spruce and other conifers developed in the dark (von Wettstein, 1958; Selstam and Widell, 1986). The light-independent protochlorophyllide reductase and the two light-dependent protochlorophyllide reductases expressed in the dark in developing conifer cotyledons make this differentiation of plastid structures understandable. The light-independent enzyme permits the synthesis of chlorophyll and the assembly of active photosystems and light-harvesting complexes in the grana (Lewandowska and Oquist, 1980), whereas a light-dependent protochlorophyllide reductase is responsible for the assembly of the prolamellar bodies. From an evolutionary point of view, cyanobacteria and algae (which neither store protochlorophyllide in the dark nor form prolamellar bodies) "invented" the light-independent and light-dependent protochlorophyllide reductases. By duplicating the gene for the latter enzyme, the seed-producing spermatophytes have evolved the prolamellar body-specific reductase.

**CHLOROPHYLL BINDING PROTEINS**

All chlorophyll molecules in the chloroplast are bound non-covalently to proteins in the photosynthetic membrane. Our present knowledge of the structure of these chlorophyll binding proteins has recently been compiled and reviewed by Jansson (1994) and Simpson and Knoetzlel (1995). In the reaction center of photosystem II (Figure 12), the D1 and D2 transmembrane polypeptides hold the "special pair" chlorophyll molecules in place by coordination with two chlorophyll a binding proteins has recently been compiled and reviewed by Jansson (1994) and Simpson and Knoetzal (1995).
histidine residues. A few additional scavenger chlorophyll molecules are also located in this protein dimer. These two polypeptides are encoded in the chloroplast genome, as are the chlorophyll a binding proteins CP47 and CP43, which comprise the inner antenna of the reaction center. These proteins each bind ~45 chlorophyll molecules and approximately six \( \beta \)-carotene molecules. In photosystem I, the dimer-forming PSIA and PSIB membrane-spanning polypeptides are encoded in the chloroplast DNA and coordinate the special pair chlorophyll P700 between them. They also bind together ~90 antenna chlorophyll a molecules and nine to 18 \( \beta \)-carotene molecules.

Both reaction center complexes are surrounded by chlorophyll a and b binding proteins (shown in light green in Figure 12). These proteins are encoded in the nucleus, synthesized on cytoplasmic ribosomes, imported into the chloroplast, and inserted into the photosynthetic membrane. Six polypeptides encoded by the \( \text{Lhcb} \) genes are associated with photosystem II, and four proteins encoded by the \( \text{Lhca} \) genes are attached to photosystem I. Most of the genes are present in one or two copies per nuclear genome, but the most abundant protein in the light-harvesting complex II of photosystem II, \( \text{Lhcb1} \), is encoded by a multigene family with three to 16 members in different plants. Their molecular weights range from 22 to 28 kD, and they contain three highly conserved helix domains and two conserved loops (cf. Jansson, 1994). Electron crystallography at 3.4 Å resolution reveals three LHCII particles as a trimeric complex in the unit cell. The individual light-harvesting polypeptides form two tilted membrane-spanning \( \alpha \)-helices (1 and 3) of unusual length (31 to 33 amino acids) and a perpendicular membrane-spanning helix of ~20 amino acids.

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**Figure 12.** Model of the Thylakoid Membrane Showing the Known Polypeptides in the Major Complexes.

The polypeptides are identified by gene and/or protein name and are color coded according to their electron transport cofactors. Light-harvesting proteins binding only chlorophyll a are shown in dark green, whereas those binding both chlorophyll a and b are shown in light green (\( \text{Lhca1-4} \) and \( \text{Lhcb1-6} \)). The “special pair” chlorophyll in photosystem II (P680) is coordinated between the \( \text{D1} \) and \( \text{D2} \) polypeptides (lightest green). That of photosystem I (P700) is ligated between the \( \text{psaA} \) and \( \text{psaB} \)-encoded polypeptides, which also bind 90 light-harvesting antenna chlorophyll a molecules.

**PSII, photosystem II.** \( \text{Q}_{\alpha} \), tightly bound quinone; \( \text{Q}_{\theta} \), exchangeable quinone; \( \text{psbE,F} \), cytochrome \( \delta_{559} \); \( \text{psbO,P,Q} \), extrinsic oxygen-evolving complex; \( \text{psbH} \) to \( \text{psbS} \), polypeptides of PSII and water-splitting enzyme. Freeze fracture membrane particles: PFs, protoplasmic fracture face of stacked membranes; LHCII, light-harvesting complex of photosystem II; EFs, ectoplasmic fracture face of stacked membranes; ESs, ectoplasmic surface face of stacked membranes.

**Cyt b5/f.** cytochrome \( \text{b}_{5} \) (red)-cytochrome \( \text{f} \) (red) complex participating in the photosynthetic electron transport between photosystem II and I and pumping protons across the membrane. pet, photosynthetic electron transport protein (chloroplast encoded); Pet (nucleus encoded); PetH, ferredoxin nucleotide reductase (FNR); PetG, polypeptide binding quinone that accepts electrons plus protons and donates electrons to the next carrier together with a release of protons as part of the proton pump across the membrane; PetE, copper-containing plastocyanin (blue); Fmu, protoplasmic fracture face of unstacked membrane.

**FNR1, ferredoxin nucleotide reductase complex for cyclic photophosphorylation.** PetF, ferredoxin.

**PSI, photosystem I.** \( \text{A}_{0} \), chlorophyll molecule; \( \text{A}_{1} \), phylloquinone; \( \text{X} \), 4Fe-4S cluster; \( \text{psaC} \), polypeptide carrying two 4Fe-4S clusters, A and B; \( \text{psaD, psaE, psaF, psaG, psaH, psaK, psaL, and psaN} \), nucleus-encoded subunits of PSI; \( \text{psaL} \) and \( \text{psaN} \), chloroplast-encoded subunits of PSI. \( \text{CF}_{1} \), extrinsic part and \( \text{CF}_{0} \), membrane-embedded part of coupling factor (ATP synthase). The \( \text{CF}_{0} \) component forms a proton channel, whereas the \( \text{CF}_{1} \) component protrudes into the stroma and couples ATP synthesis to proton translocation. The \( \alpha, \beta, \gamma \), and \( \epsilon \) subunits are encoded in chloroplast DNA, as are the channel subunits I, III, and IV. Subunits \( \gamma \) and \( \delta \) are encoded in the nucleus. PSu, protoplasmic surface face of unstacked membranes.

Updated from Simpson and von Wettstein (1989).
acids. Helices 1 and 3 bind all eight chlorophyll a and six chlorophyll b molecules, which are held in close proximity by the helices inside the photosynthetic membrane to ensure energy transfer. The helices also bind the xanthophyll lutein, and some of the polypeptides bind violaxanthin, p-carotene, and neoxanthin. These carotenoids serve as accessory pigments for absorption of blue light (see Bartley and Scolnik, 1995, this issue).

Assembly of the imported light-harvesting polypeptides and their insertion into the thylakoid membrane seem to require the presence of chlorophyll and lutein, as indicated by the following experiments. The major chlorophyll a/b binding protein can be expressed in E. coli, isolated, and folded in vitro, but only in the presence of chlorophyll a, chlorophyll b, and lutein (Paulsen et al., 1993). In barley mutants that are blocked in chlorophyll a synthesis, transcription of the structural genes for light-harvesting polypeptides proceeds at normal rates (Batschauer et al., 1986). However, the polypeptides do not accumulate in the thylakoids, probably because they are not stabilized with chlorophyll and thus are proteolytically degraded. The viable barley chlorophyll b-less mutant chlorina-f2 is deficient in LHCII, although normal levels of LHCII mRNA are present in the mutant (Batschauer et al., 1986). A limited amount of the translated and imported polypeptide binds chlorophyll a and is inserted into the thylakoids as an intrinsically unstable chlorophyll protein (Bassi et al., 1985).

It will be interesting to explore to what extent attachment of chlorophyll to the chlorophyll a/b binding proteins is required for their transport from the chloroplast envelope through the stroma into the thylakoid membrane (Nielsen et al., 1994; Robinson and Klosegen, 1994). It would also be helpful to identify mutants that are disturbed in the transport pathway. The temperature-sensitive viable mutant chlorina-104 fails to incorporate the Lhca2 and Lhcb1 light-harvesting proteins of photosystem II into the photosynthetic membrane at the restrictive temperature, but it does so when moved to the permissive temperature (Knoetzel and Simpson, 1991). Isolation of mutant plastids developed at restrictive or permissive temperature and import experiments with in vitro-transcribed and translated Lhca2 and Lhcb1 precursor proteins will show whether an import or transport component is affected in the mutant.

Insight is also being gained into chlorophyll's importance for translating mRNA for the proteins that bind chlorophyll a exclusively. These proteins are encoded in chloroplast DNA and are translated on polysomes attached to the thylakoids. In wild-type etioplasts and in plastids of mutants blocked in the synthesis of porphyrins and chlorophyll, transcription of the genes encoding D1, CP43, and CP47 of photosystem II and PSII is blocked. Translation of the mRNA-encoding reaction center polypeptide D1 revealed ribosome pausing as in the wild type, but the mRNA intermediates characteristic for pausing polypeptides were missing (Kim et al., 1994). It was concluded that the viridis-115 protein stabilizes the mRNA intermediates such that in its absence, incorporation of the polypeptides of the photosystem II core into the membrane cannot take place. In barley, the nuclear gene mutant viridis-zb lacks the reaction center complex of photosystem I but incorporates its chlorophyll a/b binding proteins and assembles a complete photosystem II (Hiller et al., 1980). This mutant provides the opportunity to analyze the assembly of photosystem I by in vitro expression of all 13 cloned subunit genes and import of the subunit precursors into the mutant plastids.

**PERSPECTIVES**

The identification, physical mapping, and nucleotide sequencing of the 45-kb photosynthesis gene cluster in Rhodobacter and closely related organisms have probably provided us with the primary structure of all genes necessary for the synthesis of chlorophyll and a basic frame of the photosynthetic apparatus. Fortunately, the primary structure of these genes is also conserved considerably in the corresponding genes of cyanobacteria, which in turn reveal sequence homology to the chlorophyll biosynthesis genes in algae and higher plants. This provides access to sufficient amounts of the enzymes and regulatory molecules required for the synthesis of the chlorophyll molecule by their expression in vitro and in heterologous hosts. Biochemical and structural analysis by spectroscopy, x-ray crystallography, and nuclear magnetic resonance spectroscopy will remain the cornerstones for elucidating the mechanisms of the individual enzymatic steps. Complementation of mutants in Rhodobacter defective in individual biosynthetic steps with proteins from higher plants and algae provides a powerful tool to identify the corresponding enzymes and their catalytic domains. The biggest challenge is to understand the regulation of chlorophyll synthesis and the assembly of the membrane proteins that hold the chlorophyll molecules in place and in close proximity for their tasks in harvesting sunlight and converting the light energy into chemical energy. Studies with isolated chloroplasts, isolated photosynthetic membranes, and isolated protein complexes will continue to provide exciting insights into the molecular mechanisms of photosynthesis.
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