

Phycobilisome Mobility and Its Role in the Regulation of Light Harvesting in Red Algae^{1[W]}

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Red algae represent an evolutionarily important group that gave rise to the whole red clade of photosynthetic organisms. They contain a unique combination of light-harvesting systems represented by a membrane-bound antenna and by phycobilisomes situated on thylakoid membrane surfaces. So far, very little has been revealed about the mobility of their phycobilisomes and the regulation of their light-harvesting system in general. Therefore, we carried out a detailed analysis of phycobilisome dynamics in several red alga strains and compared these results with the presence (or absence) of photoprotective mechanisms. Our data conclusively prove phycobilisome mobility in two model mesophilic red alga strains, *Porphyridium cruentum* and *Rhodella violacea*. In contrast, there was almost no phycobilisome mobility in the thermophilic red alga *Cyanidium caldarium* that was not caused by a decrease in lipid desaturation in this extremophile. Experimental data attributed this immobility to the strong phycobilisome-photosystem interaction that highly restricted phycobilisome movement. Variations in phycobilisome mobility reflect the different ways in which light-harvesting antennae can be regulated in mesophilic and thermophilic red algae. Fluorescence changes attributed in cyanobacteria to state transitions were observed only in mesophilic *P. cruentum* with mobile phycobilisomes, and they were absent in the extremophilic *C. caldarium* with immobile phycobilisomes. We suggest that state transitions have an important regulatory function in mesophilic red algae; however, in thermophilic red algae, this process is replaced by nonphotochemical quenching.

Photosynthetic light reactions are mediated by pigment-binding protein complexes located either inside the thylakoid membrane (e.g. chlorophyll-binding proteins of both photosystems) or associated on the membrane surface

(e.g. phycobilisomes [PBsomes] in cyanobacteria and red algae). Recent progress in structural biology has allowed the construction of high-resolution structural models of most photosynthetic protein complexes (for review, see Fromme, 2008) together with their large-scale organization into supercomplexes (for review, see Dekker and Boekema, 2005). However, the dynamics of these supercomplexes and the mobility of particular light-harvesting proteins *in vivo* are still poorly understood (for review, see Mullineaux, 2008a; Kaňa, 2013; Kirchhoff, 2014). The importance of protein mobility in various photosynthetic processes, like nonphotochemical quenching and state transitions, has been explored mostly based on indirect *in vitro* experiments, including single-particle analysis (Kouřil et al., 2005), or by biochemical methods (Betterle et al., 2009; Caffarri et al., 2009). Recent studies on the mobility of light-harvesting proteins using live-cell imaging (for review, see Mullineaux, 2008a; Kaňa, 2013) have elucidated the importance of protein mobility for photosynthetic function (Joshua and Mullineaux, 2004; Joshua et al., 2005; Goral et al., 2010, 2012; Johnson et al., 2011). In addition, the redistribution of respiratory complexes in cyanobacterial thylakoid membranes plays an essential role in controlling electron flow (Liu et al., 2012).

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It is generally accepted that the mobility of most of the transmembrane photosynthetic proteins is very restricted in the thylakoid. The typical effective diffusion coefficient of photosynthetic proteins is somewhere between 0.01 and $0.001 \mu\text{m}^{-2} \text{s}^{-1}$ (Kaňa, 2013). A similar restriction in membrane protein mobility has also been described for bacterial membranes (Dix and Verkman, 2008; Mika and Poolman, 2011). In fact, this is very different in comparison with what we know for other eukaryotic membranes (e.g. plasma membrane and endoplasmic reticulum), where membrane-protein diffusion can be faster by 1 or 2 orders of magnitude (Lippincott-Schwartz et al., 2001). Therefore, macromolecular crowding of proteins has been used to rationalize the restricted protein mobility in thylakoid membranes of chloroplasts (Kirchhoff, 2008a, 2008b). Indeed, atomic force microscopy studies have shown that there is a dense packing and interaction of complexes in the photosynthetic membranes (Liu et al., 2011). Therefore, the diffusion of photosynthetic proteins in the thylakoid membrane is rather slow, and it increases only in less crowded parts of thylakoids (Kirchhoff et al., 2013). The current model of photosynthetic protein mobility thus proposes the immobility of protein supercomplexes, such as PSII (Mullineaux et al., 1997; Kirchhoff, 2008b), with only a small mobile fraction of chlorophyll-binding proteins represented by external antennae of photosystems, including light harvesting complex of PSII in higher plants (Consoli et al., 2005; Kirchhoff et al., 2008) or iron stress-induced chlorophyll-binding protein A in cyanobacteria (Sarcina and Mullineaux, 2004).

The restricted mobility of internal membrane supercomplexes (photosystems) contrasts with the relatively mobile PBsomes (Mullineaux et al., 1997; Sarcina et al., 2001). PBsomes are sizeable biliprotein supercomplexes (5–10 MD) attached to the thylakoid membrane surface with dimensions of approximately $64 \times 42 \times 28 \text{ nm}$ (length \times width \times height; Arteni et al., 2008; Liu et al., 2008a). PBsomes are composed of chromophore-bearing phycobiliproteins and colorless linker polypeptides (Adir, 2005; Liu et al., 2005). They serve as the main light-harvesting antennae in various species, including cyanobacteria, red algae, glaucocystophytes, and cryptophytes. Although a single PBsome is composed of hundreds of biliproteins, absorbed light energy is efficiently transferred toward a specific biliprotein that functions as a terminal energy emitter (Glazer, 1989). From there, energy can be transferred to either PSI or PSII and used in photosynthesis (Mullineaux et al., 1990; Mullineaux, 1992, 1994). In typical prokaryotic cyanobacteria and eukaryotic red algae, PBsomes are composed of two main parts: (1) allophycocyanin (APC) core proteins adjacent to the thylakoid membrane; and (2) peripheral rod proteins made from phycocyanin only or from a combination of phycocyanin together with phycoerythrin. Such complex and modular composition allows for different spectroscopic properties of PBsomes and thus their complementary absorption in the spectral region that is not covered by chlorophyll-binding proteins.

PBsome mobility has been studied only in a few types of cyanobacteria (for review, see Kaňa, 2013). PBsomes have been recognized as a mobile element with an effective diffusion coefficient of about $0.03 \mu\text{m}^2 \text{s}^{-1}$ for *Synechococcus* sp. PCC 7942 (Mullineaux et al., 1997; Sarcina et al., 2001). The effective diffusion coefficient value depends on lipid composition, temperature, and the size of the PBsome (Sarcina et al., 2001). The diffusion coefficient reflects PBsome mobility, but it is not affected singularly by physical diffusion processes, and the role of PBsome-photosystem interaction is an open question (Kaňa, 2013). PBsome mobility seems to be related to the requirement of light-induced PBsome redistribution during state transitions (Joshua and Mullineaux, 2004). The mechanism of state transitions in cyanobacteria is still rather questionable (for review, see Kirilovsky et al., 2014). As PSII seems to be immobile, it has been suggested that PBsomes interact with photosystems only transiently and that physical redistribution (diffusion) of PBsomes is crucial for the state transition (Mullineaux et al., 1997). The importance of such long-distance diffusions, however, should be tested experimentally in more detail (Kaňa, 2013), as an alternative theory of the state transition proposed only slight PBsome movement (shifting) between photosystems (McConnell et al., 2002). However, in both cases, PBsome mobility (i.e. the PBsome's ability to move) is required (Kaňa, 2013).

Red algae are the eukaryotic representatives of phototrophs containing PBsomes (Su et al., 2010). They represent the ancestor of photosynthetic microorganisms from the red clade of photosynthesis (Yoon et al., 2006; Wang et al., 2013), which includes various model organisms such as diatoms, chromerids, or dinoflagellates. Red algae contain a unique combination of antennae systems on their membrane surfaces, which are formed mostly by hemispherical PBsomes (Mimuro and Kikuchi, 2003; Arteni et al., 2008). Red algae also contain transmembrane light-harvesting antennae (Vanselow et al., 2009; Neilson and Durnford, 2010; Green, 2011) associated mostly with PSI (Wolfe et al., 1994). Therefore, red algae represent a functionally important eukaryotic model organism; however, few facts are known about the regulation of its light-harvesting efficiency, although it seems to be connected with photoprotection in the reaction center (Delphin et al., 1996, 1998; Krupnik et al., 2013). The presence of photoprotective NPQ in PBsomes of prokaryotic cyanobacteria has been conclusively proven (Kirilovsky et al., 2014); however, this mechanism seems to be missing in eukaryotic phycobiliproteins of cryptophytes (Kaňa et al., 2012b) and red algae. Moreover, the presence (or absence) of PBsome mobility has not been confirmed conclusively (Liu et al., 2009).

Therefore, we carried out a detailed study of PBsome mobility in red algal chloroplasts to determine the role of mobility in the regulation of light-harvesting efficiency. We found that red alga PBsomes are a mobile protein complex with effective diffusion coefficient between 2.7×10^{-3} and $13 \times 10^{-3} \mu\text{m}^2 \text{s}^{-1}$ in all studied mesophilic strains. It contrasted with PBsomes in extremophilic red algal strains (*Cyanidium caldarium*), where

PBsome mobility under physiological conditions was highly restricted (effective diffusion coefficient of approximately $0.6 \times 10^{-3} \mu\text{m}^{-2} \text{s}^{-1}$). The restriction of PBsome mobility in extremophilic *C. caldarium* was due to a tight interaction of PBsomes with both photosystems and not to changes in lipid desaturation, an effect typical for extremophiles. The PBsome-photosystem interaction was weakened for *C. caldarium* grown at suboptimal temperatures, resulting in a pronounced increase in PBsome mobility thanks to PBsome decoupling from the photosystem. This result shows that PBsome mobility in this strain is limited by the strength of the PBsome-photosystem interaction rather than by the restriction of diffusion by factors such as macromolecular crowding. Moreover, our study allows us to describe two different models of light-harvesting antenna regulation in red algae. In mesophilic strains (*Porphyridium cruentum* and *Rhodella violacea*), absorbed light is redistributed between photosystems in a process of state transition that requires PBsome mobility. On the contrary, in extremophilic *C. caldarium*, PBsome are strongly coupled to photosystems and excess light is dissipated by a process of nonphotochemical quenching, as has been described recently (Krupnik et al., 2013).

RESULTS

PBsome Diffusion in Red Algae: Fluorescence Recovery after Photobleaching Images

The mobility of red alga PBsomes was detected by the fluorescence recovery after photobleaching (FRAP)

method, which allows direct measurement of photosynthetic protein mobility (for review, see Kaňa, 2013). A 633-nm laser was used to excite PBsomes, and their emission was measured above 665 nm. The recovery of the fluorescence signal during the FRAP measurements reflected mostly PBsome mobility and not the mobility of chlorophyll-binding proteins with an emission in this range (Supplemental Fig. S1) because (1) the 633-nm excitation induces chlorophyll emission through PBsomes attached to photosystems, so PBsome mobility is required to see fluorescence recovery after bleaching; and (2) there is almost no mobility of chlorophyll-binding proteins (Supplemental Fig. S2), so chlorophyll-binding proteins cannot participate in the fluorescence recovery seen in Figure 1. Figure 1 thus represents a typical FRAP image sequence showing PBsome mobility in *P. cruentum*. Images were made every 8 s before and after the application of high-intensity bleach. The prebleach image (top left in Fig. 1) shows a typical distribution of PBsome fluorescence in a single cell of *P. cruentum*. Application of high laser intensity across the *x* axis (red rectangle in Fig. 1) reduced PBsome fluorescence to about 40% of the initial value. The observed fluorescence recovery in the bleached zone (yellow circle in Fig. 1) showed PBsome mobility in this red alga.

The kinetics of recovery of the PBsome fluorescence shown in Figure 1 was further analyzed numerically (Fig. 2). We quantified fluorescence changes during FRAP measurements by pixel averaging in part of the bleached area (yellow circle in Fig. 1). The plot confirmed the recovery of PBsome fluorescence in the place

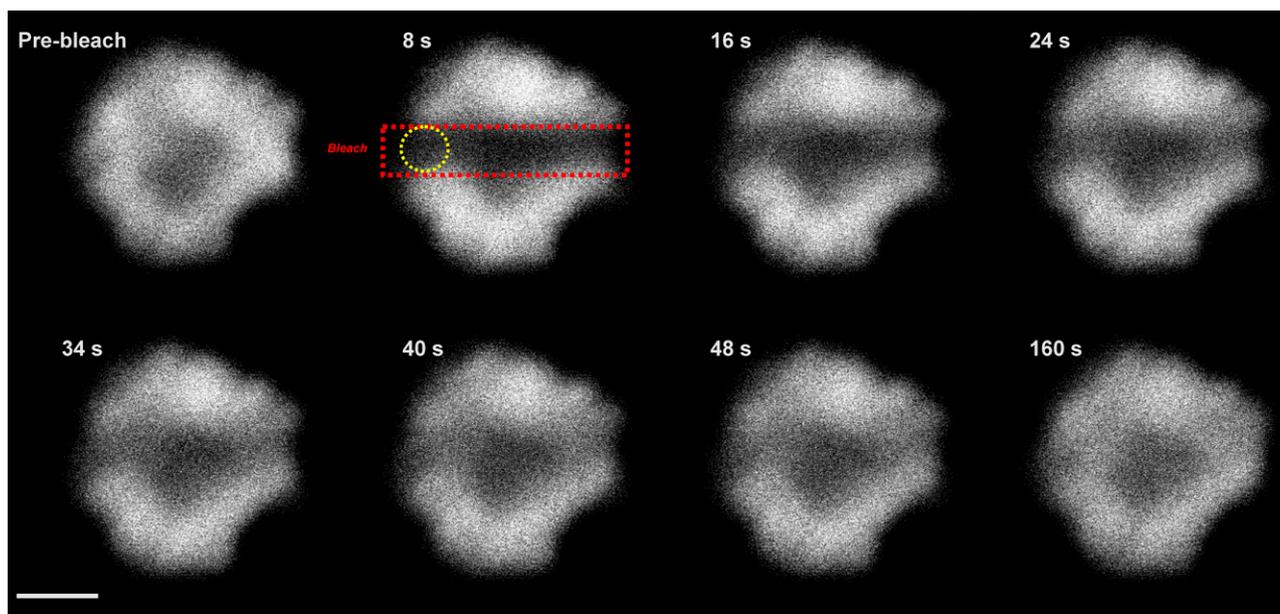


Figure 1. Representative FRAP image sequence for a single cell of *P. cruentum* for PBsome fluorescence. Fluorescence was excited at 633 nm, and emission was detected in the spectral range above 665 nm. First, a fluorescence image before bleaching was detected (Pre-bleach), and then the PBsome fluorescence was bleached out across the middle of the cell in the X direction (red rectangle); bleach depth (the extent of fluorescence decrease due to bleach application) was about 40% of the initial fluorescence intensity. Fluorescence in the middle of the membrane was used for further numerical analysis of the recovery in PBsome fluorescence (yellow circle). Time after bleaching is shown. Bar = 3 μm .

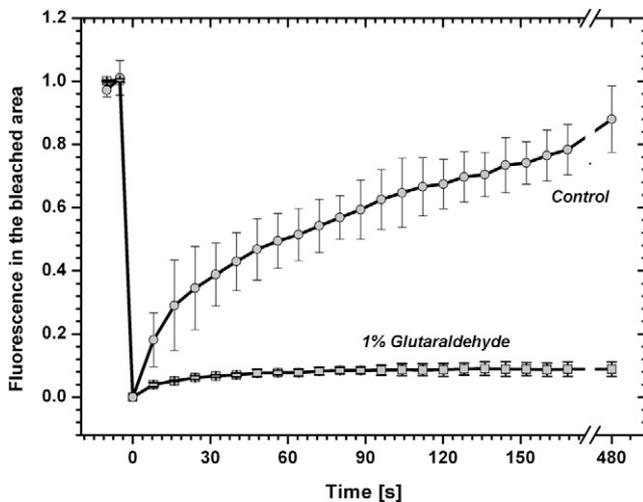


Figure 2. Time course of PBsome fluorescence recovery at the center of the bleached thylakoid of *P. cruentum*. The FRAP routine was measured for a control cell or with 1% glutaraldehyde, which cross links proteins and stops protein diffusion. Data were calculated from the FRAP image sequence presented in Figure 1; values are relative to fluorescence prior to bleaching, and the typical bleach depth before normalization was about 40% of the initial fluorescence. Data represent average values and *sd* for *n* = 9 with three biological replications.

of bleaching to about 80% of the initial value after 8 min of recovery. To exclude the possible effect of internal recovery in PBsome fluorescence due to the recovery of chromophore dark states (for review, see Kaňa, 2013), the FRAP routine was measured also for cells treated with 1% glutaraldehyde, which cross links proteins and stops protein mobility. In this case, we found only a very small recovery of fluorescence after bleaching (about 8%), which probably reflected the previously described internal fluorescence recovery in PBsomes (Liu et al., 2009). However, glutaraldehyde-insensitive recovery represented a dominant fraction of fluorescence recovery after photobleaching. Our data clearly show the presence of PBsome mobility and the minimal effect of internal fluorescence recovery in PBsomes of *P. cruentum*.

Properties of PBsome Diffusion in Red Algae

PBsome mobility was further studied by detailed analysis of bleach profiles (Fig. 3). The bleach profile represents a one-dimensional projection of fluorescence changes across the *x* axis in a selected area of the thylakoid (red rectangle in Fig. 3A and projection results in Fig. 3B), which can be used for the calculation of the effective diffusion coefficient (see “Materials and Methods”). Comparison of the prebleach and 8-s profiles (Fig. 3B) showed a decrease in PBsome fluorescence intensity (bleaching profile) due to the application of high-intensity laser power. The application of high-intensity bleach reduced total fluorescence to about 40% of the initial value due to the destruction of a portion of the phycobilin pigments (fluorescence intensity in the central bleached zone in Fig. 3A). The central zone was flanked by areas

that showed a slight fluorescence increase immediately after bleaching (zone D, 1.5 μm , in Fig. 3, A and C). While the PBsome fluorescence in the central zone (central zone in Fig. 3A) slowly recovered with time after bleaching (16, 32, and 120 s in Fig. 3B), fluorescence intensity out of the bleached area (especially in zone D, 1.5 μm , in Fig. 3, A and C) was decreasing with time after bleaching. This loss of fluorescence out of the central zone (described as fluorescence loss in photobleaching; for review, see Kaňa, 2013) represents an additional proof of the diffusional exchange of nonfluorescent PBsomes from the central (bleached) zone with fluorescent PBsomes from the outer nonbleached zones. The exchange of fluorescent and nonfluorescent PBsomes is also visible in the detailed numerical analysis of fluorescence intensity in different distances from the middle of the bleached area (Fig. 3C). PBsome fluorescence in the central zone increased during the time-lapse experiment, in contrast with areas out of the central zone (1.5 and 1 μm in Fig. 3, A and C). Interestingly, at a distance of about 0.75 μm from the central zone, the movement of fluorescent PBsomes into and out of the 0.75- μm zone was equilibrated, resulting in a constant PBsome fluorescence during the whole time-lapse experiment (0.75 μm in Fig. 3C). These results provide a clear indication of PBsome mobility in *P. cruentum*.

Species Variability in PBsome Diffusion in Red Algae

We tested PBsome mobility in a mesophilic red alga with typical red algal hemispherical PBsomes, *P. cruentum* (Koller et al., 1977), and in *R. violacea*, which contains hemidiscoidal PBsomes that are more typical for cyanobacteria. The FRAP measurements proved that both of these types of PBsomes are mobile in mesophilic strains. Hemispherical PBsomes were significantly slower in their mobility (*P. cruentum* in Fig. 4) in comparison with much faster hemidiscoidal PBsomes (*R. violacea* in Fig. 4). In addition, the PBsome mobility was also measured in the extremophilic red alga *C. caldarium* (Fig. 4), which was isolated from highly acidic hot springs. Interestingly, PBsome mobility was almost undetectable (Fig. 4) in this extremophilic red alga grown at its optimal temperature at 38°C. Cultivation of *C. caldarium* at suboptimal temperatures (18°C) resulted in PBsome mobility (Fig. 4). However, this PBsome mobilization was accompanied by a very slow cell growth rate (Fig. 5), as the suboptimal temperature greatly increased doubling time. Therefore, the increase in PBsome mobility for *C. caldarium* from 18°C (Fig. 4) did not improve the vitality and physiology in these extremophilic red algae.

We further analyzed the extent of the PBsome mobile fraction in particular species. The steady-state value of fluorescence recovery at 480 s after bleaching was between 20% and 90% (Fig. 4). The internal effect of fluorescence recovery in PBsomes (diffusion-uncoupled fluorescence recovery; Kaňa, 2013) was estimated for all tested strains by FRAP measurements with

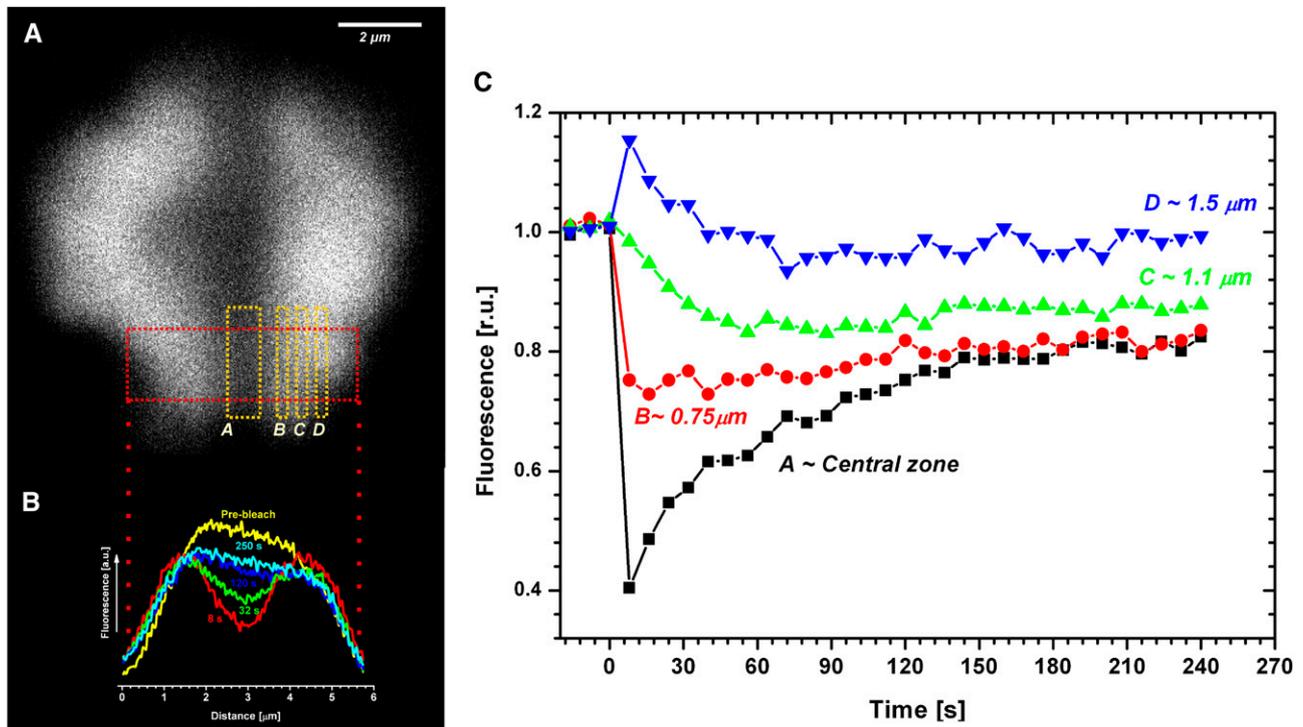


Figure 3. Detailed analysis of the bleach profiles during the measurement of PBsome mobility in the red alga *P. cruentum*. A, Representative image taken 8 s after bleaching. Areas (zones) with different distances from the central zone (A) are labeled by yellow rectangles as B ($0.5 \mu\text{m}$), C ($1.1 \mu\text{m}$), and D ($1.5 \mu\text{m}$). B, Typical dynamics of a one-dimensional bleach profile taken at characteristic times after bleaching (as labeled). Curves were constructed based on two-dimensional images at different time during the FRAP routine (Fig. 1) by averaging of two-dimensional data across x axes (the red curve 8 s represents averaging across the red rectangle in A). C, Dynamic changes in average values of the A to D zones presented in A during the FRAP routine (Fig. 1). r.u., Relative units.

1% glutaraldehyde, which can cross link proteins and stop protein diffusion (data not shown). Nevertheless, it represented only a minimal fraction of recovery, as in all tested strains it was below 10% (data not shown). The PBsome mobile fraction corrected for this internal effect is shown in Table I. Our analysis conclusively showed a relatively high mobile fraction of PBsomes in mesophilic red algae (79.2% and 88.9% for *P. cruentum* and *R. violacea*, respectively) and only a minimal mobile fraction of PBsomes in the extremophilic red alga *C. caldarium* (19.8%) at optimal growth conditions (38°C).

Effective diffusion coefficients of these mobile PBsomes were calculated numerically from the kinetics of bleach profiles (for details, see “Materials and Methods”). This analysis (Table I) showed significant variability in effective diffusion coefficients, with faster hemidiscoidal (*R. violacea*) and slower hemispherical (*P. cruentum*) PBsomes (between 2.7×10^{-3} and $13 \times 10^{-3} \mu\text{m}^{-2} \text{s}^{-1}$; Table I). It again confirmed the 10 times slower mobility of hemispherical PBsomes from red algae in comparison with hemidiscoidal PBsomes from the cyanobacterium *Synechococcus* sp. PCC 7942 (approximately $30 \times 10^{-3} \mu\text{m}^{-2} \text{s}^{-1}$; Mullineaux et al., 1997; Sarcina et al., 2001). It shows that hemispherical PBsomes have slower mobility in comparison with hemidiscoidal PBsomes.

In contrast with mesophilic species with diffusion coefficients between 2.7×10^{-3} and $13 \times 10^{-3} \mu\text{m}^{-2} \text{s}^{-1}$, PBsome mobility in the thermophilic *C. caldarium* grown at 38°C was 1 order of magnitude slower (around $0.57 \times 10^{-3} \mu\text{m}^{-2} \text{s}^{-1}$; Table I). We were able to stimulate PBsome diffusion in *C. caldarium* only by the cultivation of this strain at suboptimal temperatures (18°C), which had the effect of limiting growth rate (Fig. 5). In this case, PBsomes could move with a diffusion coefficient around $3.84 \times 10^{-3} \mu\text{m}^{-2} \text{s}^{-1}$ (Table I). We assume that the low diffusion coefficient value for PBsomes in *C. caldarium* grown at 38°C may be related to a stronger interaction of these PBsomes with photosystems, as there was only a minimal mobile fraction (Table I). This model has been tested by 77 K fluorescence spectroscopy (see below).

77 K Fluorescence Spectroscopy

Functional interaction was tested by means of 77 K fluorescence emission spectra by exciting PBsomes at 580 nm (Fig. 6). The 77 K fluorescence emission spectra showed typical bands (Fig. 6). We compared experimental spectra with the spectra detected from isolated protein complexes of both *P. cruentum* (Gantt et al.,

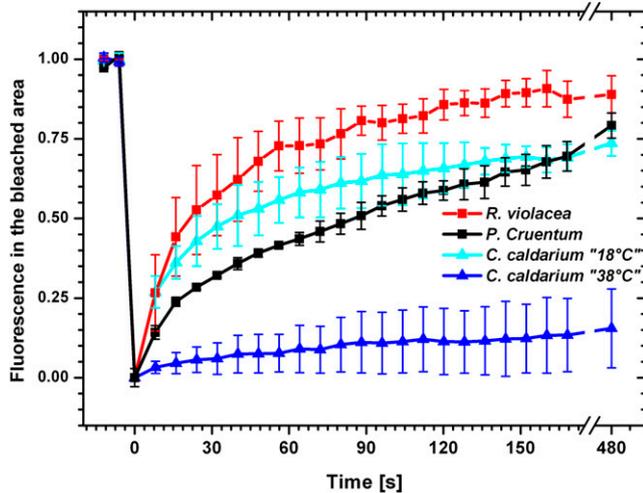


Figure 4. Time courses of fluorescence recovery for PBsome fluorescence in different red algae species, *P. cruentum*, *R. violacea*, and *C. caldarium*, grown at two temperatures, suboptimal 18°C (*C. caldarium* 18°C) and optimal 38°C (*C. caldarium* 38°C). Fluorescence values are relative to fluorescence prior to bleaching and were normalized to the changes in fluorescence in the nonbleached area. The effect of internal recovery was corrected by subtraction of FRAP curves measured with 1% glutaraldehyde (Fig. 2). Data represents averages and SD for $n = 12$.

1979; Bumba et al., 2004) and *C. caldarium* (Gardian et al., 2007; Busch et al., 2010). This allowed us to interpret the observed emission maxima as follows: (1) fluorescence emission at 644 nm in *P. cruentum* (F644 in Fig. 6A) was due to R-phycocyanin fluorescence emission (Ley et al., 1977), and emission at 650 nm in *C. caldarium* (F650 in Fig. 6B) originated from C-phycocyanin; (2) fluorescence emission around 661 nm (F661 in Fig. 6C) represented APC fluorescence; (3) fluorescence maxima around 684 nm (F684 in Fig. 6) represented a mixture of chlorophyll *a* fluorescence from PSII and APC-B emission of a terminal PBsome emitter (Ley et al., 1977); (4) fluorescence emission around 694 nm (F694, F695) is emitted from PSII; and (5) far-red light emission forms in *P. cruentum* (F708 in Fig. 6A) and in *C. caldarium* (F728 in Fig. 6B) reflected the fluorescence of red chlorophylls from PSI and its antennae. The 77 K fluorescence emission spectra for excitation of PBsomes (at 580 nm) were further used as a rough estimate of energy transfer from PBsomes to PSI (reflected by fluorescence emissions F708 and F728) or to PSII (reflected by F695 emission). The significant emission from both photosystems following excitation to PBsomes clearly showed PBsome energy flow to PSI and PSII.

We also studied the PBsome interaction with photosystems in *C. caldarium* grown at 18°C, which showed a higher mobile fraction of PBsomes (Table I). The F695-F728 ratio for excitation to PBsomes at 580 nm was the same for *C. caldarium* grown at 18°C and 38°C (Fig. 6C), so it did not change during adaptation to low temperature. The adaptation to the suboptimal temperature also did not affect the PSI-PSII ratio, as can be deduced

from the F695-F728 ratio with excitation of chlorophylls at 434 nm, which was around 0.06 for *C. caldarium* from both growth temperatures of 38°C and 18°C (Fig. 6B). There was only a small decrease in the relative amount of PBsome in comparison with chlorophyll (compare PBsome absorbance at approximately 625 nm and chlorophyll at approximately 675 nm in Supplemental Fig. 3, A and B and Supplemental Table S1) between *C. caldarium* grown at 18°C and 38°C. The dominant change we observed during adaptation to low-temperature stimulation of PBsome mobility was a relative increase in PBsome fluorescence maxima between 650 and 684 nm (Fig. 6C), C-phycocyanin fluorescence (F650), and APC fluorescence (F662 and F684). Such an increase in PBsome fluorescence indicates a higher fraction of PBsomes that are energetically uncoupled from photosystems in *C. caldarium* grown at 18°C in comparison with cells grown at 38°C; we suggest that this is the reason for the higher mobile fraction of PBsomes in these cells (Table I). Therefore, the immobility of PBsomes in *C. caldarium* grown at 38°C is caused by a strong PBsome interaction with photosystems. As *C. caldarium* grew much faster at 38°C than at 18°C (Fig. 5), this indicates that the strong PBsome interaction with photosystems and the almost complete lack of PBsome mobility (Table I) represent an optimal physiological state in these thermophilic red alga allowing a faster growth rate. This contrasts with mesophilic red algae (*P. cruentum* and *R. violacea*), where PBsomes are present as fast mobile protein supercomplexes (Table I).

Composition of Lipids and PBsome Mobility

The interaction of PBsomes with photosystems is affected by lipid composition, especially by fatty acid saturation (Sarcina et al., 2001). Therefore, we quantified the extent of fatty acid saturation in two representatives

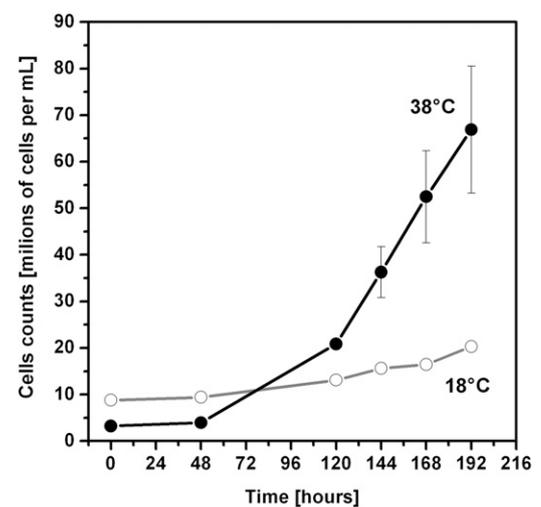


Figure 5. Growth curves of *C. caldarium* cells at 18°C or 38°C. Adapted cells from the stationary phase were diluted (to less than 10^7 cells mL⁻¹) and counted every 1 to 2 d by Multisizer 3.

Table I. PBsome mobile fraction and effective diffusion coefficients

Effective diffusion coefficient (D) was calculated from experimental FRAP data (see “Materials and Methods”). The presented mobile fraction represents fluorescence recovery 480 s after bleaching, where internal recovery of fluorescence unconnected with diffusion (recovery measured with glutaraldehyde) was subtracted. Data represent averages and SD for $n = 12$.

Parameter	<i>P. cruentum</i>	<i>R. violacea</i>	<i>C. caldarium</i> 38°C	<i>C. caldarium</i> 18°C
Mobile fraction (%)	79.2 ± 10.6	88.9 ± 5.7	19.8 ± 5.3	73.7 ± 7.8
D ($\times 10^{-3} \mu\text{m}^{-2} \text{s}^{-1}$)	2.69 ± 0.77	13.04 ± 9.15	0.57 ± 0.18	3.84 ± 2.19

red algae with mobile (*P. cruentum*) and immobile (*C. caldarium*) PBsomes grown at 18°C and 38°C. Isolated membranes of all three strains were analyzed by gas chromatography (see “Materials and Methods”). We detected typical fatty acids in various ratios (Table II). Four fatty acids (saturated C16:0 [palmitic acid] and C18:0 [stearic acid] and unsaturated C18:1n9 [oleic acid] and C18:2n6 [linoleic acid]) were detected in high amounts (above 1% from total lipid content) in all studied strains. There were few species-specific fatty acids (asterisks in Table II): highly unsaturated fatty acids detected only in *P. cruentum* (C18:3n6 [γ -linolenic acid], C20:4n6 [arachidonic acid], and C20:5n3 [eicosapentaenoic acid]) and fatty acids observed only in *C. caldarium* (C14:0 [myristic acid] and C18:3n3 [α -linolenic acid]). This indicates that mesophilic and thermophilic red algae adapt to different living conditions by changes in their membrane fatty acid composition; this is in line with previous studies done for *P. cruentum* (Khozin et al., 1997; Khozin-Goldberg et al., 2000) and *C. caldarium* (Allen et al., 1970).

The average extent of fatty acid unsaturation was characterized by the double bond index (DBI), which reflects membrane fluidity, as it represents a relative amount of double bonds in membrane lipids. The highest fatty acid saturation (lowest DBI) was observed in thermophilic *C. caldarium* cells with limited PBsome mobility; this is in line with the reduction in membrane fluidity (i.e. higher lipid saturation) usually observed during growth at higher temperatures. The low DBI observed in *C. caldarium* grown at 38°C increased only slightly during adaptation to low growth temperature (from 1.2 to 1.3; Table II). On the contrary, the membranes of the mesophilic red alga *P. cruentum* with mobile PBsomes (Table I) were more fluid, as shown by higher DBI (Table II). Therefore, higher PBsome mobility can be observed in more fluid membranes with more unsaturated lipids (i.e. with higher DBI). The effective diffusion coefficient and PBsome mobile fraction were higher (Table I) in more fluid membranes of mesophilic *P. cruentum* (higher DBI in Table II) and smaller in extremophilic *C. caldarium* grown at 38°C (Table I), which had less fluid membrane (lower DBI in Table II). However, the correlation between higher PBsome mobility and higher membrane fluidity was not general, as *C. caldarium* grown at 18°C had much higher PBsome mobility (Table I) without any significant increase in membrane fluidity based on DBI (Table II). There was also no change in the fatty acid chain length, another determinant for membrane fluidity, between *C. caldarium* cultures grown at 18°C and 38°C

(Table II). Therefore, the stimulation of PBsome mobility in *C. caldarium* grown at 18°C in comparison with *C. caldarium* grown at 38°C is not caused by membrane fluidization due to lipid desaturation; rather, it is caused by a weakening of the photosystem-PBsome interaction,

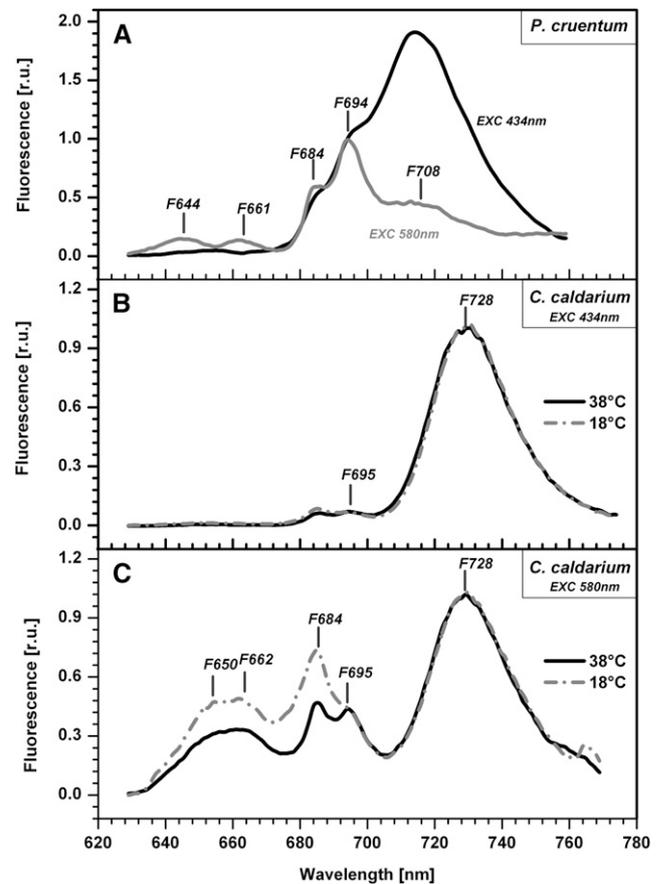


Figure 6. 77 K fluorescence emission spectra of *P. cruentum* and *C. caldarium*. A, 77 K fluorescence emission spectra of *P. cruentum* grown in optimal temperature (18°C) detected for excitation to PBsomes (at 580 nm; gray curve) or to chlorophyll *a* (at 434 nm; black curve). B, 77 K fluorescence emission spectra of *C. caldarium* grown in optimal (38°C) and suboptimal (18°C) temperatures that were detected for excitation to chlorophyll *a* (at 434 nm). C, 77 K fluorescence emission spectra of *C. caldarium* grown in optimal (38°C) and suboptimal (18°C) temperatures that were detected for excitation to PBsomes (at 580 nm). Data represent typical curves for dark-adapted cells normalized to 694 nm (A) or 728 nm (B and C). The positions of characteristic peaks are marked. r.u., Relative units.

Table II. Fatty acid composition of red algae membranes

Values represent DBI, average fatty acid chain length (FA chain), and relative amounts (in %) of the following fatty acids: C14:0, myristic acid; C16:0, palmitic acid; C16:1, palmitoleic acid; C18:0, stearic acid; C18:1n7, cis-7-octadecenoic acid; C18:1n9, oleic acid; C18:2n6, linoleic acid; C18:3n3, α -linolenic acid; C18:3n6, γ -linolenic acid; C20:3n6, dihomo- γ -linolenic acid; C20:4n6, arachidonic acid; and C20:5n3, eicosapentaenoic acid. Data represent average values and SD for $n = 3$. Asterisks indicate fatty acids that are specific either for extremophilic or mesophilic red algae. n.d., Not detectable.

Parameter	<i>P. cruentum</i>	<i>C. caldarium</i>	
		18°C	38°C
DBI	2.5 ± 0.2	1.3 ± 0.1	1.2 ± 0
FA chain	18.20 ± 0.05	17.47 ± 0.05	17.36 ± 0.01
C14:0	n.d.	0.40 ± 0.18*	0.26 ± 0.01*
C16:0	34.34 ± 0.94	26.69 ± 1.17	31.99 ± 0.15
C16:1	2.13 ± 0.36	0.46 ± 0.08	0.23 ± 0.07
C18:0	1.7 ± 0.8	4.77 ± 0.50	3.54 ± 0.25
C18:1n7	2.51 ± 1.53	0.55 ± 0.06	0.13 ± 0.03
C18:1n9	2.97 ± 2.5	6.43 ± 2.49	8.59 ± 0.04
C18:2n6	6.45 ± 0.72	58.89 ± 4.03	54.31 ± 0.55
C18:3n3	n.d.	0.56 ± 0.32*	0.18 ± 0.15*
C18:3n6	0.31 ± 0.05*	n.d.	n.d.
C20:3n6	0.55 ± 0.03	1.24 ± 0.18	0.78 ± 0.06
C20:4n6	16.24 ± 1.78*	n.d.	n.d.
C20:5n3	32.11 ± 3.41*	n.d.	n.d.

as indicated by the 77 K fluorescence spectra (Fig. 6). *C. caldarium* grown at suboptimal 18°C had a higher fraction of PBsomes uncoupled from photosystems (increase in PBsome fluorescence in Fig. 6C) that do not contribute to light harvesting.

Physiological Importance of PBsome Mobility

The presence of PBsome mobility in cyanobacteria has already been correlated with the occurrence of state transitions (Joshua and Mullineaux, 2004). It is a process that equilibrates absorbed energy distribution between photosystems (for review, see Kirilovsky et al., 2014). State transitions in cyanobacteria are typified by an increase in maximal fluorescence during the dark-light transition, as cyanobacteria are in the low-fluorescence state II in the dark. We observed a similar effect for dark-adapted *P. cruentum* exposed to low blue light but not in extremophile *C. caldarium* (Supplemental Fig. S4). This is, to our knowledge, the first indication showing the presence of state transitions in mesophilic red algae and its absence in extremophilic red algae. The process of state transition was further studied by the measurement of fluorescence changes in dark adapted cells during exposure to high orange light (Fig. 7). The fast light-induced fluorescence increase to its maximum (in hundreds of milliseconds) can be followed by another much slower fluorescence rise (in tens of seconds) from the plateau called S to the second M peak. Therefore, it is called the S-M rise. The S-M rise is dominant in cyanobacteria in contrast to the higher plants and green algae (for review, see Papageorgiou et al., 2007). The S-M

rise was recently recognized as a result of the state 2-to-state 1 transition in cyanobacteria (Kaňa et al., 2012a). The slow fluorescence S-M rise in maximal fluorescence was detected also in *P. cruentum* between 10 and 50 s after irradiation of dark-adapted cells (Fig. 7A). The fluorescence increase was accelerated by the presence of 3-(3,4-dichlorophenyl)-1,1-dimethylurea, which amplified the state 2-to-state 1 transition due to a lack of plastoquinone pool reduction. On the contrary, the S-M fluorescence rise was fully stopped by 1% glutaraldehyde, which cross links proteins and also stops PBsome mobility measured from FRAP (Fig. 2). Therefore, we suggest that the presence of PBsome mobility in red algae correlates with state transition-induced changes in fluorescence in high-intensity orange light.

Indeed, the slow S-M fluorescence rise was totally missing in *C. caldarium* grown at optimal temperature (Fig. 7B), which also did not show any PBsome mobility. Moreover, there was no S-M fluorescence rise for *C. caldarium* from suboptimal growth temperatures (data not shown). This shows that an increase in PBsome mobility for *C. caldarium* grown at 18°C is not reflected in the appearance of state transitions. PBsomes in *C. caldarium* grown at 18°C are more weakly bound to photosystems, in contrast to *C. caldarium* grown in 38°C (compare the PBsome diffusion coefficient in Table I). The absence of the S-M rise in *C. caldarium* from 18°C also confirms that the higher PBsome diffusion coefficient observed at these conditions ($3.84 \times 10^{-3} \mu\text{m}^{-2} \text{s}^{-1}$; Table I) is caused by a simple PBsome decoupling from photosystems and is not related to any physiological process of light energy

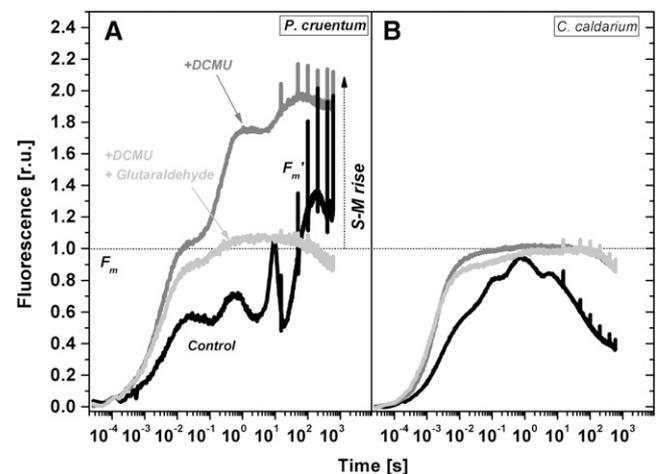


Figure 7. Time course of chlorophyll a fluorescence measured with *P. cruentum* (A) and *C. caldarium* (B). Samples without any addition (control) or in the presence (where indicated) of 10 μM 3-(3,4-dichlorophenyl)-1,1-dimethylurea (DCMU) or 1% glutaraldehyde are presented. Data are normalized to the maximal fluorescence obtained in the dark (F_m) before the fluorescence time course was recorded. Several saturation pulses were applied to obtain maximum fluorescence in the light (F_m'). The characteristic slow fluorescence rise (S-M rise) reflecting the state 2-to-state 1 transition is marked. For fluorescence excitation, orange diodes ($\lambda_e = 590 \text{ nm}$, half-width $\Delta\lambda = \pm 20 \text{ nm}$) were used. r.u., Relative units.

distribution between photosystems. Therefore, PBsome mobility in *C. caldarium* is limited mostly by the strength of PBsome-photosystem interaction and not by a physical diffusion process. The state transition connected with PBsome redistribution, therefore, does not play such an important role in extremophilic red algae like *C. caldarium*. Rather, this species regulates energy arriving to PSII by some other mechanism, like nonphotochemical quenching in the reaction center of the extramophilic red alga *Cyanidioschyzon merolae* (Krupnik et al., 2013), or by excitation energy spillover between photosystems (Kowalczyk et al., 2013).

DISCUSSION

In this article, we show that PBsomes in mesophilic red algae are mobile light-harvesting antennae (Fig. 4), in line with PBsomes from prokaryotic cyanobacteria (for review, see Kaňa, 2013). By contrast, PBsomes in extremophilic red alga from hot acidic springs (*C. caldarium*) are almost immobile (Fig. 4; Table I), similar to the immobile phycobiliproteins in the lumen of cryptophytes (Kaňa et al., 2009b). The immobility of *C. caldarium* PBsomes is caused by their strong interaction with photosystems. This interaction is weaker in *C. caldarium* grown at suboptimal temperatures (18°C), resulting in PBsome decoupling from photosystems and a higher mobility (Fig. 4; Table I). Such PBsome decoupling from photosystems has been proposed to serve as one of the protoprotective mechanisms in cyanobacteria (Kaňa et al., 2009a; Tamary et al., 2012; for review, see Kirilovsky et al., 2014). In *C. caldarium*, PBsome decoupling from low growth temperature (18°C) causes increases in PBsome mobility measured by FRAP (Fig. 4; Table I). The increase in PBsome mobility was not accompanied by an increase in growth rate (Fig. 5), and it was also not involved in the stimulation of state transitions (Fig. 7). On the other hand, it could represent a photoprotective mechanism in extremophilic red algae to redirect excessive PBsome-absorbed irradiation away from the PBsomes (Kirilovsky et al., 2014).

We also observed a correlation between the presence of PBsome mobility and state transitions (measured from fluorescence) for PBsomes in mesophilic red algae (*P. cruentum*). In fact, the application of glutaraldehyde, cross linking PBsomes with photosystems, inhibits both processes, state transitions (Fig. 7) and PBsome mobility (Fig. 2). In comparison with cyanobacterial PBsomes in *Synechococcus* sp. PCC 7942 (Mullineaux et al., 1997), the effective diffusion coefficient for red algae PBsome has been found to be about 10 and three times less in *P. cruentum* and *R. violacea*, respectively (Table I). One possible explanation could be a different structure and organization of hemidisoidal PBsomes in *Synechococcus* sp. PCC 7942 and hemispherical PBsomes from *P. cruentum*. In fact, PBsomes are represented by three distinct classes: (1) primitive rod-like PBsomes present in *Acaryochloris marina* (Theiss et al., 2011), in *Gloeobacter violaceus* (Krogmann et al., 2007), a common rock-dwelling

organism (Mareš et al., 2013), and a similar rod-like PBsome with CpcG2 linker found in *Synechocystis* sp. PCC 6803 (Kondo et al., 2007); (2) widely distributed hemidisoidal PBsomes are typical for cyanobacteria (e.g. *Synechococcus* sp. PCC 7942 and *Synechocystis* sp. PCC 6803) and for some red algae (e.g. *R. violacea*; Koller et al., 1977); and (3) hemispherical (hemiellipsoidal) types of PBsomes specific for most of the red algae, such as *P. cruentum* (Arteni et al., 2008). Our study demonstrates a reduced mobility of hemispherical PBsomes in comparison with hemidisoidal ones (Fig. 4). However, the mobility of hemidisoidal PBsomes in *R. violacea* is still slower in comparison with their analogs from *Synechococcus* sp. PCC 7942, with a diffusion coefficient of approximately $30 \times 10^{-3} \mu\text{m}^{-2} \text{s}^{-1}$ (Mullineaux et al., 1997; Sarcina et al., 2001). Therefore, PBsome mobility is not only affected by PBsome structure and size (Sarcina et al., 2001), and other factors also should be addressed in future experiments. The different PBsome mobilities in *P. cruentum* and *R. violacea* also could be caused by variations in the PBsome-photosystem ratio. To check this, we estimated the photosystem-PBsome ratio by means of deconvolution of absorption spectra (Supplemental Fig. S3; Supplemental Table S1). However, there was no significant difference in the photosystem-PBsome ratio (A_{440}/A_{630} in Supplemental Table S1), indicating that differences in this ratio are not the cause of the observed differences in PBsome mobility (Fig. 4). Indeed, the PBsome-photosystem ratio seems to be maintained constant over a wide range of growth conditions (Cunningham et al., 1989, 1990).

The slower PBsome mobility in red algae (in comparison with cyanobacteria) may be caused by PBsomes packing on thylakoid membrane surface, as reported recently for *P. cruentum* (Liu et al., 2008a). Such a crowded arrangement may seriously restrict PBsome diffusion due to the presence of physical obstacles, such as PBsomes (Liu et al., 2008a). This is similar to the effect that has already been found in the mobility of light-harvesting antennae in the overcrowded grana of higher plants (Kirchhoff, 2007). Another difference between cyanobacteria and red algae is the presence of intrinsic transmembrane light-harvesting antennae in red algae that are missing in cyanobacteria. It is plausible that the interaction of PBsomes with these antennae could slow PBsome mobility in red algae. The lower PBsome mobility in red algae in comparison with cyanobacteria could be the result of stronger PBsome-photosystem interaction, as the FRAP method reflects not only diffusional movement but also protein-protein interactions (Kaňa, 2013). The mobility of the relatively large PBsomes also could be affected by the limited width of the interthylakoidal space where the PBsome is situated. The flexibility of this interthylakoidal space might be another factor affecting changes in PBsome mobility. It was shown recently that the width of the interthylakoidal space can vary between light and dark (Nagy et al., 2012; Liberton et al., 2013). The importance of light-induced adjustment of the multilamellar thylakoid membrane system for the mobility of PBsomes and

other thylakoid proteins remains an open question that should be tested in future experiments.

Besides the mobile PBsomes, we also detected a small immobile fraction of PBsomes in mesophilic red algae (Table I). The immobile fraction was dominated in the extremophilic red alga *C. caldarium* under physiological conditions (*C. caldarium* 38°C in Fig. 4). The mobile fraction of PBsomes in *P. cruentum* (about 80%; Table I) was smaller in comparison with the cyanobacterium *Synechococcus* sp. PCC 7942, where roughly 100% of PBsomes are mobile (Mullineaux et al., 1997). This raises the question of the origin of this immobile fraction of PBsomes. This immobile fraction of PBsomes could be caused by the heterogeneity of the PBsome interaction with photosystems, as they act as an external antenna of both photosystems in cyanobacteria (Mullineaux, 1994; Ashby and Mullineaux, 1999; Rakhimberdieva et al., 2001) and also in red algae (Stadnichuk et al., 2011; Yokono et al., 2011). Moreover, there is a different organization of PSII in cyanobacteria (dimers) and red algae (tetrameric) in the arrangement of PSII complexes (Lange et al., 1990; Tsekos et al., 2004), which could explain the presence of an immobile fraction of PBsomes. The partial immobility of red alga PBsomes may be due to the presence of special proteins, called regulators of phycobilisome association with photosystems (the so-called Rpa-like proteins), that have already been described for cyanobacteria (Emlyn-Jones et al., 1999; Ashby et al., 2002; Joshua and Mullineaux, 2005). One of them, RpaC, a small transmembrane protein, has been recognized as a critical factor for state transitions, although its presence in red algae is questionable. Earlier biochemical studies suggested a role for one of the allophycocyan subunits, ApcE (sometimes called Lcm, for linker core membrane), in the PBsome-photosystem interaction (for review, see Liu et al., 2005; Mullineaux, 2008b). However, even though this protein is essential for the stabilization of the PBsome core, its direct role in PBsome binding to photosystems is still questionable (Ajilani and Vernotte, 1998).

The immobile PBsomes were dominant in the extremophilic red alga *C. caldarium*. Our data show the crucial role of the strength of the PBsome-photosystem interaction for the mobility/immobility of PBsomes in this organism. It has been suggested previously that the PBsome interaction with photosystems is rather weak and unstable; therefore, all PBsomes were considered to be uniformly mobile (Mullineaux et al., 1997; Sarcina et al., 2001). However, our results indicate that this assumption requires more direct experimental data in order to be confirmed. In the case of *C. caldarium*, the PBsome-photosystem interaction was weakened in *C. caldarium* grown at low temperatures (18°C) and was rather strong in *C. caldarium* grown at 38°C (Fig. 4; Table I). This has been deduced from the increase/decrease in PBsome fluorescence at 77 K, reflecting the lower/higher efficiency of energy transfer from PBsomes to photosystems (Fig. 6C). It resulted in the acceleration of the PBsome diffusion of *C. caldarium* from low temperatures and also in an increase of the mobile fraction of PBsomes

from 20% to 74% (Table I). Since we have excluded the role of higher lipid desaturation in PBsome decoupling from photosystems (desaturation changed only slightly between 18°C and 38°C cultures of *C. caldarium*; Table II; Fig. 7), we suggest that the immobility of PBsomes in the extremophilic *C. caldarium* at optimal growth temperature is caused by a strong interaction of PBsomes with photosystems or with their antennae.

The firm interaction of PBsomes with PSI has been already shown in the extremophilic red alga *C. merolae*, a close relative of *C. caldarium*, where PSI together with membrane-extrinsic PBsome rods and integral membrane light-harvesting antennae form a very stable supercomplex (Busch et al., 2010). Moreover, such a tight coupling between antennae proteins in extremophilic red algae has already been found between PSI and its light-harvesting antennae (Thangaraj et al., 2011). This suggests that the stronger interaction between light-harvesting proteins in extremophilic red algae seems to be a consequence of their adaptation to high temperatures (above 40°C).

We also tried to determine the physiological importance of PBsome diffusion in red algae. In cyanobacteria, the presence of PBsome mobility has been correlated with the presence of state transitions (Joshua and Mullineaux, 2004) that act in the redistribution of absorbed light between PSI and PSII. Several mechanisms of state transitions have been suggested for cyanobacteria (for review, see Kirilovsky et al., 2014), and many of them propose PBsome displacement or at least slight PBsome rearrangement (McConnell et al., 2002; Joshua and Mullineaux, 2004; Kirilovsky et al., 2014). As photosystems and chlorophyll-binding proteins in cyanobacteria (Mullineaux et al., 1997) and in red algae (Supplemental Fig. S2) are almost immobile, the importance of PBsome mobility for the process of state transitions has already been suggested for cyanobacteria (Joshua and Mullineaux, 2004). Our data have revealed a similar correlation between PBsome mobility and state transitions in red algae. The correlation (between PBsome mobility and state transitions) does not necessarily mean the requirement of a long-distance PBsome diffusion for state changes. It only shows the necessity of some PBsome mobility for state changes. A limiting step for this mobility could be either a PBsome displacement (diffusion) or PBsome-photosystem interactions (Kaňa, 2013). The particular contribution and importance of those two processes need to be tested experimentally. These experiments are of special importance especially in light of the recent finding that PSI, PSII, and PBsomes can form a supercomplex (Liu et al., 2013) where PBsomes do not have to be moved far in order to induce a change in excitation flow from PBsomes to PSI or PSII.

It seems that in extremophilic red algae such as *C. caldarium*, state transitions are not so important in regulating light energy transfer between PBsomes and photosystems; the absence of state transitions correlates with the PBsome immobility in *C. caldarium* (Fig. 4). As a consequence, these extremophiles have developed other mechanisms to regulate energy arriving to reaction

centers: nonphotochemical quenching (fluorescence decrease in Fig. 7). Indeed, we recently conclusively proved the presence of a reaction center type of non-photochemical quenching in the extremophilic red alga *C. merolae* (Krupnik et al., 2013).

All these results allow us to define two simplified models of light-harvesting antenna regulation in red algae with respect to PBsome (im)mobility. In the first mechanism, typical for mesophilic strains (*P. cruentum* and *R. violacea*), absorbed light can be redistributed between photosystems in a process of state transitions; therefore, PBsome mobility is relatively high. On the contrary, in extremophilic *C. caldarium*, PBsomes are strongly coupled to photosystems and excess light must be dissipated by a process on nonphotochemical quenching (Krupnik et al., 2013) or by PBsome decoupling (Liu et al. 2008b, Kirilovsky et al. 2014). An overlap between this model and other photoprotective mechanisms, like energy spillover between photosystems (Kowalczyk et al., 2013), is a matter for future research.

MATERIALS AND METHODS

Cell Growth and Sample Preparation

The mesophilic marine red alga *Porphyridium cruentum* (UTEX B637) was cultivated in adapted Porphyridium medium (Brody and Emerson, 1959), mesophilic *Rhodella violacea* was grown in an artificial seawater medium with f/2 nutrient addition (plus soil extract), and extremophilic *Cyanidium caldarium* was grown in Cyanidium medium, pH 2.5 (Allen, 1959). Low light intensity was used (approximately $30 \mu\text{mol m}^{-2} \text{s}^{-1}$, with a day/night cycle of 12 h/12 h), and cells were continuously bubbled with air (Kaňa et al., 2012b). Temperature was set either to 18°C (*R. violacea* and *P. cruentum*) or 38°C (*C. caldarium*).

FRAP Measurements and Data Analysis

FRAP measurements of PBsomes were performed with a laser-scanning confocal microscope (Nikon PCM2000) equipped with a red helium-neon laser (633-nm line) and with a 60× oil-immersion lens of numerical aperture (NA) 1.4 and an Olympus FV1000 equipped with a red helium-neon laser (633-nm line) with a 100× oil-immersion UPLSAPO lens of NA 1.4 and a sampling speed 2 μs per pixel. The mobility of chlorophyll-binding proteins was measured with a laser-scanning confocal microscope (Nikon PCM2000) with a 60× oil-immersion lens of NA 1.4; fluorescence was excited by an argon laser (488 nm) and detected with a Schott RG665 red glass filter (transmitting above 665 nm). All measurements were performed at room temperature (20°C). A dichroic mirror transmitting above 650 nm (Nikon PCM2000) and a dichroic mirror with the edge at 635 nm (DM635; Olympus) were used to separate excitation and fluorescence light. The PBsome emission for measurements with the Nikon PCM2000 was selected with a Schott RG665 red glass filter (transmitting above 665 nm) and detected for a low laser power (approximately 3%). The PBsome emission for measurements with the Olympus FV1000 was selected using the variable band-pass filter system (100 nm above 655 nm).

The bleach line across the cell was induced with full red helium-neon laser power (for about 3 s) by fast switching the confocal microscope to X-scanning (Nikon PCM2000) or by the application of 100% intensity to a selected rectangular area of the cell (FV1000). The bleach intensity was set to reach about 40% bleach depth. Postbleach images were recorded typically every 8 s for 4 min with low laser power (10%). The series of FRAP images were analyzed with the Olympus FluorViewer 10-ASW software and with the public domain Java image-processing program ImageJ 1.45d (Abramoff et al., 2004). A one-dimensional bleaching profile was obtained by integration across the cell in the X direction in the parallel direction to the bleach line (Mullineaux et al., 1997). The baseline fluorescence from the unbleached cell was subtracted; the recovery curves were normalized to fluorescence changes in nonbleached cells, and double normalization including acquisition bleaching correction (Phair et al., 2004) was used during data processing.

Calculation of Effective Diffusion Coefficients from FRAP Results

Effective diffusion coefficients were calculated numerically based on a recently published method (Papáček et al., 2013). One-dimensional bleach profiles across x axes were constructed from experimental data exported from the FluorViewer 10-ASW software (Olympus) and further transformed in a two-dimensional data set using ImageJ 1.45d (Abramoff et al., 2004). A data set representing one-dimensional postbleach profiles was imported into the numerical software and used for effective diffusion coefficient calculation by a numerical routine without regularization (Papáček et al., 2013). The applied numerical algorithm is based on an algorithm that minimized the objective function representing differences between experimental data and numerical simulation. The numerical scheme was constructed based on the finite difference approximation of a one-dimensional Fickian diffusion equation. Dirichlet boundary conditions were applied for effective diffusion coefficient by numerical simulation of eight postbleach profiles.

The objective function was minimized for all eight postbleach profiles independently (Kaňa et al., 2013). The minimization of the objective function represents a one-dimensional optimization problem that was solved by the trust-region method from an open-source system of the universal functional optimization methods, a series of algorithms useful for solving optimization problems (Lukšan et al., 2013). The applied numerical routine (version FRAP 2.0) computed effective diffusion coefficients iteratively using the following parameters: (1) one-dimensional postbleach profiles were normalized to the same data taken from a prebleach image; (2) the first postbleach profiles were taken as initial conditions; (3) the central part of the bleach and surrounding areas were selected for analysis with an average size of around 2 μm ; (4) data noise from all experimental data sets were reduced by smoothing using Fourier transformation; (5) the first eight postbleach profiles were used for numerical analysis; and (6) the Crank-Nicholson implicit scheme was used for the computation of simulated profiles (Papáček et al., 2013).

Lipid Extraction and Analysis

Cells were harvested at the exponential growth phase, collected biomass was centrifuged, and the pellet was diluted by resuspension in solution (800 μL of water, 2 mL of pure methanol, and 1 mL of dichloromethane [DCM]) and sonicated in an ultrasonic bath for 10 min at room temperature. The solution was diluted again in 1 mL of water and 1 mL of DCM and briefly vortexed. Phase separation of lipids in a glass tube with DCM was achieved by centrifugation (500g, 10 min, and 4°C), the lower DCM layer was removed and placed into clean evaporation vials with 2 mL of DCM, and the vortexing and centrifugation steps were repeated two more times. Lower phases were pooled and dried using a rotary evaporator. Dried extracts were redissolved in DCM:methanol (2:1, v/v) at a concentration of 1 mg of lipid extract per 20 μL and stored at -70°C for further analysis.

The lipid extract was methylated as described before (Kainz et al., 2002). An aliquot of lipid extract (approximately 0.5 mg) in DCM:methanol (2:1, v/v) was added to 15 μg of heptadecanoic acid and dried in a stream of N_2 . The sample was diluted by mixing with 0.5 mL of hexane and 1 mL of 10% (w/w) borontrifluoride-methanol (Supelco, Sigma-Aldrich) and placed into a heating bath set to 85°C for 1 h. A total of 0.5 mL of chloroform-extracted water and 2 mL of hexane were added, and the sample was vortexed and centrifuged at 500g for 5 min. The upper hexane layer was removed to another vial for evaporation. The remaining mixture was washed two times with 2 mL of hexane as described above. Layers were pooled and concentrated under a stream of nitrogen to a final volume of 20 μL . Methylated samples (1 μL) were used for further quantitative and qualitative analysis on a GC-FID-HRGC 5300 device (Carlo Erba) equipped with a TR-FAME column (30 m \times 0.32 mm, 0.25 μm film thickness; Thermo Fisher). Helium was used as the carrier gas at a pressure of 200 kPa. The temperature program was as follows: initial temperature of 140°C, temperature increase after injection at a rate of 4°C min^{-1} up to 240°C, and then constant temperature (240°C) for 10 min. The retention times of fatty acid methyl esters were compared with known standards (Supelco 37 Component FAME Mix and Supelco PUFA No. 3 [Supelco, Sigma-Aldrich]). Amounts of individual fatty acids were calculated as described before (Masood et al., 2005). The DBI was calculated as the sum of the relative contents of saturated fatty acids, each multiplied by the number of its double bonds. Fatty acid chain length was calculated as the sum of the relative contents of fatty acids, each multiplied by number of carbons in its fatty acid chain.

Fluorescence Measurements

Fluorescence emission spectra at 77 K for dark-adapted cells were measured with an Aminco-Bowman Series 2 spectrofluorometer (Thermo Electron). The sample was infiltrated on membrane filters that were fitted in a sample holder and immersed in an optical Dewar flask filled with liquid nitrogen. Possible effects of fluorescence reabsorption were tested for every sample by measuring spectra with different amounts of sample added to the filter and checking that the F695-F30 ratio was constant. Excitation was provided at 434 and 580 nm, each with a 4-nm bandwidth. Fluorescence emission was scanned with a 2-nm bandwidth between 630 and 800 nm. The experimental data were deconvoluted using Origin Pro 9.1 Peak Analyzer (OriginLab) using Gaussian curves. The positions of proposed peak maxima were restricted to a 4-nm range around the proposed maxima, and all other parameters were set to be free for the minimization procedure, driven by χ^2 (with 10^{-6} precision).

Chlorophyll *a* fluorescence induction kinetics were measured with the FL-100 Fluorometer (Photon Systems Instruments) as described previously (Kaňa et al., 2012a). Orange actinic light (590 nm; half-width $\Delta\lambda = 20$ nm; $300 \mu\text{mol m}^{-2} \text{s}^{-1}$) was used for excitation, and fluorescence induction was measured in the 690- to 750-nm range. Samples were dark adapted for 20 min before measurements, and the maximal fluorescence of closed PSII reaction centers was measured during a saturation flash (590 nm; length, 200 ms; approximately $1,200 \mu\text{mol photons m}^{-2} \text{s}^{-1}$) in the dark. Subsequently, variable fluorescence kinetics, induced by orange actinic irradiation, were measured over a 10-min interval with logarithmically increasing intervals between the measured points. Fluorescence emission spectra were deconvoluted by Gaussian peaks in OriginPro version 9.1 (OriginLab) as described (Kaňa et al., 2009a).

Absorption Spectra

Absorption spectra were measured with a spectrophotometer (Unicam UV 550; Thermo Spectronic) equipped with an integrating sphere. Cells were collected on nitrocellulose membrane filters (Pragochema), and the filters were positioned in the integrating sphere. Absorbance was measured between 400 and 800 nm, with a bandwidth of 4 nm. Absorption spectra were deconvoluted by Gaussian peaks in OriginPro version 9.1 (OriginLab) as described (Kaňa et al., 2009a).

Physiological Measurements

The growth rate of *C. caldarium* was estimated using cell counts. For the estimation of growth rate at different temperatures, *C. caldarium* cells were adapted for 3 weeks to a given temperature (18°C or 38°C) before measurements. The adapted cells from the stationary phase were diluted (to less than 10^7 cells mL^{-1}) and counted every 1 to 2 d using a Multisizer 3 (Beckmann Coulter) with a 50- μm aperture in Isotone II (Beckmann Coulter).

Supplemental Data

The following materials are available in the online version of this article.

Supplemental Figure S1. Room temperature fluorescence emission spectra of the red alga strain *P. cruentum* for excitation to PBsomes at 615 nm.

Supplemental Figure S2. Mobility of chlorophyll-binding proteins of *P. cruentum*.

Supplemental Figure S3. Absorption spectra of *C. caldarium*, *P. cruentum*, and *R. violacea*.

Supplemental Figure S4. Changes in variable fluorescence in dark-adapted cells of *P. cruentum* and *C. caldarium* exposed to low blue light.

Supplemental Table S1. Results of the deconvolution of absorbance spectra presented in Supplemental Figure S3, with focus on chlorophyll (CHL - $A_{440\text{nm}}$) and phycocyanin + allophycocyanin (PC+APC - $A_{630\text{nm}}$) bands.

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