Cystathionine β-Synthase (CBS) Domains Confer Multiple Forms of Mg\(^{2+}\)-dependent Cooperativity to Family II Pyrophosphatases

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**Background:** Enzymes and transporters may contain regulatory nucleotide-binding cystathionine β-synthase domains.

**Results:** Both substrate and adenine nucleotides bind cooperatively to four bacterial cystathionine β-synthase domain-containing pyrophosphatases.

**Conclusion:** Cystathionine β-synthase domains internally inhibit catalysis and impart cooperativity; both effects are modulated by bound nucleotides.

**Significance:** The observed regulatory complexity may be characteristic of cystathionine β-synthase domain-containing proteins involved in hereditary diseases.

Regulated family II pyrophosphatases (CBS-PPases) contain a nucleotide-binding insert comprising a pair of cystathionine β-synthase (CBS) domains, termed a Bateman module. By binding with high affinity to the CBS domains, AMP and ADP usually inhibit the enzyme, whereas ATP activates it. Here, we demonstrate that AMP, ADP, and ATP bind in a positively cooperative manner to CBS-PPases from four bacteria: Desulfitobacterium hafniense, Clostridium novyi, Clostridium perfringens, and Eggertella lenta. Enzyme interaction with substrate as characterized by the Michaelis constant (K\(_m\)) also exhibited positive catalytic cooperativity that decreased in magnitude upon nucleotide binding. The degree of both types of cooperativity increased with increasing concentration of the cofactor Mg\(^{2+}\) except for the C. novyi PPase where Mg\(^{2+}\) produced the opposite effect on kinetic cooperativity. Further exceptions from these general rules were ADP binding to C. novyi PPase and AMP binding to E. lenta PPase, neither of which had any effect on activity. A genetically engineered deletion variant of D. hafniense PPase lacking the regulatory insert was fully active but differed from the wild-type enzyme in that it was insensitive to nucleotides and bound substrate non-cooperatively and with a smaller K\(_m\) value. These results indicate that the regulatory insert acts as an internal inhibitor and confers dual positive cooperativity to CBS domain-containing PPases, making them highly sensitive regulators of the PP\(_i\) level in response to the changes in cell energy status that control adenine nucleotide distribution. These regulatory features may be common among other CBS domain-containing proteins.

Pyrophosphate (PP\(_i\)) is formed as a by-product of nearly 200 biosynthetic reactions in which nucleoside triphosphates are converted to nucleoside monophosphates and PP\(_i\) (1). Many of these PP\(_i\)-generating reactions are inhibited by PP\(_i\), highlighting the importance of mechanisms that strictly control the free cellular concentration of PP\(_i\). This control is generally achieved by the hydrolysis of PP\(_i\) to phosphate via the action of specific enzymes, soluble inorganic pyrophosphatases (PPases\(^5\); EC 3.6.1.1), belonging to three non-homologous families (2–4).

Of these PPases, family II soluble PPases, found in prokaryotes, are the most active (k\(_\text{cat} = \sim 10^5 \text{ s}^{-1}\)) and structurally diverse. These homodimeric enzymes are Mn\(^{2+}\) or Co\(^{2+}\) metalloproteins but additionally require Mg\(^{2+}\) for catalysis (5), which occurs by direct attack of a hydroxide ion coordinated with three metal ions (6). Each subunit is formed by two domains connected by a flexible linker with the active site located between the domains (7, 8), making the enzyme ideally suited for allosteric regulation. Indeed, a quarter of the nearly 500 putative family II PPases sequences generally contain a 250-residue insert comprising a pair of cystathionine β-synthase (CBS) domains (Bateman module (9)), which have regulatory functions in several other proteins (10, 11–14), and a DRTGG domain of unknown function (see Fig. 1). CBS domains are quite common in nature and are found, for instance, in 75 human proteins and eight proteins of *Escherichia coli*. Mutations in CBS domains of human enzymes and membrane channels are associated with hereditary diseases, including homocystinuria, retinitis pigmentosa, Bartter syndrome, osteopetrosis, and others (13). CBS domains are self-sufficient units (see Fig. 1B) that retain structure after being removed from functional cores. Moreover, some regulatory

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\(^4\) The abbreviations used are: PPase, pyrophosphatase; AP\(_{i}\), A\(_5^\prime\), 5\(^\prime\)-P\(_4^\prime\)-diadenosine tetraphosphate; CBS, cystathionine β-synthase; cnPPase, C. novyi pyrophosphatase; cpPPase, C. perfringens pyrophosphatase; dhPPase, D. hafniense pyrophosphatase; dhP3Pase, d3CDDC, dhP3Pase devoid of regulatory part; ePPase, E. lenta pyrophosphatase; mtP3Pase, Moorella thermoacetica pyrophosphatase; TEV, tobacco etch virus; Tes, 2-(2-hydroxy-1,1-bis[hydroxymethyl])ethylamino)ethanesulfonic acid.

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Nucleotide-regulated Soluble Pyrophosphatases

proteins consist of only a Bateman module (15, 16). The majority of CBS domain–containing proteins form homodimers with the CBS domains participating in subunit interface interactions. Consequently, the two CBS domain pairs form a dislike structure (10). Interestingly, PPase appears to be the only Bateman module–containing protein in which a DRTGG domain intercalates the CBS domains (see Fig. 1). However, in rare cases, for example Moorella thermoacetica PPase (mtPPase), the enzyme contains only CBS domains in the regulatory part of the molecule. Typically, CBS domains bind adenine nucleotides, such as S-adenosylmethionine, AMP, ADP, and ATP, as regulatory ligands (10, 11–14). Each CBS domain contains a potential binding cavity for nucleotides; however, with rare exceptions (16, 17), each pair of CBS domains generally binds only one adenylate ligand. The structural and kinetic basis for regulation through CBS domains is still unknown for any protein, and CBS domain-containing PPases represent a good model enzyme for exploring this general phenomenon.

We reported previously that mtPPase (18) and the CBS domain–containing PPase of Clostridium perfringens (cpPPase) (19) are highly sensitive to micromolar concentrations of adenine nucleotides. Remarkably, AMP and ADP acted as inhibitors, whereas ATP and AP4A were activators. We also determined the three-dimensional structures of an isolated regulatory insert of cpPPase comprising two CBS domains and one DRTGG domain in complexes with AMP and AP4A (19). Rates of AMP-induced conformational transitions were much faster in cpPPase (20) than in mtPPase (21), suggesting a role for the DRTGG domain in regulatory site performance.

Here, we show that the regulatory part of the molecule, comprising two CBS domains with or without an intercalating DRTGG domain, binds adenine nucleotides cooperatively and confers catalytic cooperativity to cpPPase and three newly isolated family II PPases of Desulfitobacterium hafniense (dhPPase), Clostridium novyi (cnPPase), and Eggerthella lenta (elPPase). We found that both types of cooperativity depend on Mg2+ concentration and the identity of the bound nucleotide, further expanding our understanding of the mechanisms involved in regulating cellular PPi level. These findings were corroborated by studies of a genetically engineered dhPPase deletion variant corresponding to the isolated catalytic part of the enzyme. CBS domain–containing PPases are thus the first known oligomeric PPases whose subunits exhibit cooperativ behavior.

EXPERIMENTAL PROCEDURES

Cloning—Genomic DNA extracted from D. hafniense, C. novyi, and E. lenta (DSM numbers 10664, 14992, and 2243, respectively) was obtained from DSMZ (Deutsche Sammlung von Mikroorganismen und Zellkulturen GmbH). The sequences of full-length dhPPase (residues 1–544; YP_002458003), cnPPase (residues 1–548; YP_878001), and elPPase (residues 1–455; YP_003181314) were amplified by PCR and ligated into NdeI and XhoI restriction sites of the pET28bTEV plasmid (Novagen) for expression in E. coli strain BL21(DE3)-RIL (Stratagene) by cultivating at 37 °C in Terrific Broth medium supplemented with 30 μg/ml each of kanamycin and chloramphenicol. Gene expression was induced by incubating with 1 mM isopropyl β-thiogalactopyranoside for 3 h. Collected cells were suspended in lysis buffer (50 mM Tris−HCl, 500 mM NaCl, 20 mM imidazole−HCl, pH 7.8) and disrupted with an MSE 100-watt ultrasonic disintegrator. The cell lysate was applied to a 2-ml TALON Superflow gravity column (GE Healthcare), and dhPPase was eluted using a buffer containing 50 mM Tris−HCl, 500 mM NaCl, and 0.2 mM imidazole−HCl (pH 7.8). Imidazole was removed from eluate fractions containing dhPPase by passing through a HiPrep 26/10 column (GE Healthcare) pre-equilibrated with buffer consisting of 50 mM Tris−HCl (pH 7.8) and 500 mM NaCl. The His6 tag was removed by digesting overnight with tobacco etch virus (TEV) protease at 20 °C as described by the manufacturer after which the mixture was passed through a TALON Superflow gravity column to remove His6-containing peptides from the sample. The flowthrough was further purified by size exclusion chromatography on a Superdex 200 26/60 (GE Healthcare) column pre-equilibrated with 50 mM MOPS−KOH (pH 7.2), 150 mM KCl, 0.1 mM CoCl2, and 20 μM diethylthiinemipentaacetate. cnPPase and elPPase were expressed and purified similarly except that the His6 tag was not removed, and the buffer in size exclusion chromatography was 25 mM MOPS−KOH (pH 7.2), 50 mM KCl, and 0.1 mM CoCl2. cpPPase was expressed and purified as described previously (19). Protein solutions were stored at −40 to −80 °C.

The purity of the isolated proteins was assessed by SDS-PAGE using Phast system 8–25% gradient gels (GE Healthcare). Protein concentrations were determined with a NanoDrop spectrophotometer (Thermo Scientific) using A280 optical density values of 0.478 for dhPPase, 0.419 for dhPPaseΔCDC, 0.548 for cnPPase, 0.426 for cpPPase, and 0.493 for elPPase calculated from the corresponding amino acid compositions with ProtParam. Molar concentrations were calculated on the basis of the subunit molecular masses of 60.4, 34.5, 63.6, 60.8, and 52.5 kDa, respectively.

Kinetic Assays—The activity assay medium contained 140 μM PPi, (yielding 50 μM MgPPi, complex), 5 mM MgCl2, and 0.1 M Tes-KOH (pH 7.2) except where specified otherwise. In measurements done at higher Mg2+ concentrations, buffer concentration was decreased appropriately to maintain constant ionic strength. The reaction was initiated by adding enzyme, and Pi accumulation due to PPi hydrolysis was continuously recorded for 2–3 min at 25 °C using an automatic P2 analyzer (22). The analyzer utilizes the principle of color change.
of methyl green in the presence of phosphomolybdate and consists of a three-channel peristaltic pump and a flow-through photometer. The pump delivers a constant flow of sample, acid molybdate, and methyl green solutions, which are successively combined and directed to the flow photometer. The sensitivity of the analyzer was varied to yield full-scale recorder values of 4 or 20 μM P_i using an appropriate choice of pump tubing diameters; corresponding sample consumption rates were 5 and 1 ml/min. The dead time of the analyzer (i.e. the time from feeding the sample into the analyzer to mixing with the acid molybdate solution, which quenches the enzymatic reaction) was 10–20 s in the standard mode. The precision of initial velocity estimates for reactions yielding nonlinear time courses of P_i production was increased by decreasing the dead time to 1 s using an appropriate modification of the inlet system (23). The initial velocity values obtained in replicate measurements agreed within 5–10% and were strictly proportional to enzyme concentration.

Calculations and Data Analysis—The values of the apparent dissociation constants for the various metal ion complexes used to calculate the free metal ion concentrations at pH 7.2 were as follows: MgPP_i, 112 μM (24); Mg_2PP_i, 2.84 mM (24); MgAMP, 10.5 mM (25); MgADP, 0.42 mM (25); and MgATP, 0.034 mM (25). The values for adenine nucleotides were derived from published values of the corresponding pH-independent constants (25). The merit of using 4K_{N1}/K_{N2} instead of h to characterize cooperativity is that the former parameter is directly related to the free energy change associated with cooperativity, that is the effect of a ligand binding at one site on the binding affinity of the other site (27).

Alternatively, nucleotide binding data were fit to the Hill equation (Equation 4) to determine the Hill coefficient h.

$$A = A_0 + (A_N - A_0)/(1 + b[N]^h)$$

(Eq. 3)

Cooperative kinetics of substrate hydrolysis was analyzed in terms of Scheme 2, which assumes different Michaelis constants (K_{m1} and K_{m2}) and equal k_cat values for two active sites in the dimer (E_2); [E_0] is total enzyme concentration. The values of K_{m1}, K_{m2}, and k_cat were obtained by fitting Equation 5 to the dependence of hydrolysis rate (v) on substrate (MgPP_i) concentration at fixed Mg^{2+} concentrations.

$$v = k_{cat}[E_0](1 + 0.5K_{m2}/[S])/(1 + K_{m2}/[S] + K_{m1}K_{m2}/[S]^2)$$

(Eq. 5)

As was the case for Equation 2, the ratio 4K_{m1}/K_{m2} can be used to recognize catalytic cooperativity: its value is greater than 1 for positive cooperativity and less than 1 for negative cooperativity (26). The applicability of the microscopic/macroscopic constant concept to Michaelis constants results from the formal similarity of the Michaelis-Menten equation to the equilibrium binding equation (28) despite the fact that they are not equilibrium substrate binding constants in a general case.

The dependence of K_N on Mg^{2+} (M) concentration was fit to Equation 6 derived for Scheme 3 where (K_N)_0 and (K_N)_M are the limiting values of K_N at zero and infinite Mg^{2+} concentrations, and K_M is the metal binding constant. Equations analogous to Equation 6 were used to analyze the Mg^{2+} concentration dependence of K_m.

$$K_N = (K_N)_0(1 + [M]/K_M)/(1 + (K_N)_0[M]/(K_M)_0K_M)$$

(Eq. 6)
Nucleotide-regulated Soluble Pyrophosphatases

\[
\begin{align*}
E & \quad K_M \\
\downarrow (K_M)_0 & \quad \uparrow (K_M)_M \\
EN & \quad ENM
\end{align*}
\]

SCHEME 3. Modulation of nucleotide binding to CBS-PPase by Mg\(^{2+}\).

**RESULTS**

**Production of PPases**—The genes for CBS-PPases and the deletion variant of \(dh\)PPase (\(dh\)PPase\(\Delta CDC\); Fig. 1A) were expressed in \(E. coli\), and the proteins were purified with a yield of \(-10\) mg of pure protein/liter of medium. Constructs were designed to encode an additional N-terminal MGSSHHHHHH-\(HENLYFQGKH\) sequence containing a His\(_6\) tag to facilitate isolation by immobilized metal ion affinity chromatography. TEV protease cleaves this sequence between the Gln and Gly residues, leaving only a dipeptide extension on the protein. His\(_6\) tag removal did not affect PPase activity but diminished the aggregation of full-size \(dh\)PPase during purification and concentration procedures. Tag removal was not performed with \(dh\)PPase\(\Delta CDC\) and other PPases as it did not improve their solubility. Inactive aggregates were separated from soluble active proteins during size exclusion chromatography. The final products were at least 95% pure as estimated by SDS-PAGE.

The final purification step (Superdex 200 chromatography) was conducted in the presence of 0.1 mM Co\(^{2+}\) to stabilize the enzymes; Co\(^{2+}\) was also routinely added to enzyme stock solutions. The stabilizing effect of Co\(^{2+}\) is consistent with data showing that “canonical” family II PPases contain one site that tightly binds transition metal ions (Co\(^{2+}\) or Mn\(^{2+}\)) in addition to several more loosely binding sites with broader specificity that are apparently filled with Mg\(^{2+}\) \textit{in vivo} (5, 29, 30). All of these sites have a role in catalysis, but the tightly bound transition metal ion also has a structural role (7, 8) as it does in many other metalloenzymes. The activities of CBS-PPases preincubated with 0.1 mM Co\(^{2+}\) were somewhat higher than those incubated with Mn\(^{2+}\). Therefore, Co\(^{2+}\) was routinely used as the transition metal cofactor in these studies.

**Cooperativity of Substrate Hydrolysis by CBS-PPases**—The dependence of CBS-PPase activity on substrate concentration in the presence of 5 mM free Mg\(^{2+}\) exhibited systematic deviations from simple Michaelis-Menten kinetics as illustrated in Fig. 2 for \(dh\)PPase. The Hill coefficient \(h\), derived from this dependence using Equation 4 by setting \(A_0\) to zero, was 1.37 ± 0.02 (Fig. 2), indicating positive catalytic cooperativity. Therefore, the dependence was analyzed using Equation 5 derived for Scheme 2, which assumes different Michaelis constants \((K_{m1}^{-1} = K_{m2}^{-1} = 0)\) for two active sites. This analysis markedly increased the quality of the fit compared with the Michaelis-Menten equation (Fig. 2) as indicated by a ~12-fold lower sum of the squares of residuals. The values of \(K_{m1}^{-1}\) and \(K_{m2}^{-1}\) were found to be 26 ± 1 and 10 ± 1 \(\mu M\), respectively; the ratio \(4K_{m1}^{-1}/K_{m2}^{-1}\) (equal to 10) was thus much greater than 1, confirming positive catalytic cooperativity. Importantly, product formation curves were linear, and reaction rates were strictly proportional to enzyme concentration in all cases, ruling out the possibility that the deviation from simple kinetics seen in Fig. 2 resulted from a
dependence on Mg\(^{2+}\) the parameters (values of \(K_m\) and \(K_c\)) of the enzyme identities are indicated on the top of the panels. Values of \(k_{cat}\) were fit to the equation \(k_{cat} = k_{cat,0} + (k_{cat,0} - k_{cat,M})/\left[1 + (K_m(M)]\right)^n\) (with \(n\) equal to 1 and 2 for \(cp\) and \(cp\)PPase, respectively) where \(k_{cat,0}\) and \(k_{cat,M}\) are the limiting values of \(k_{cat}\) at zero and infinite Mg\(^{2+}\) concentrations, respectively, and \(K_m\) is the metal binding constant. The lines for \(K_m\) and \(K_c\), show the best fits for Equation 6. The horizontal dotted lines (\(h = 1\)) mark the boundary between positive and negative cooperativity. \(K_m\) values are measured in terms of the MgPPi complex. Error bars represent S.E.

FIGURE 3. Dependence of kinetic cooperativity on Mg\(^{2+}\) concentration in four CBS-PPases. The panels show (from top to bottom) the catalytic constant \(k_{cat}\), the Michaelis constants \(K_{cat}\) and \(K_c\), and the Hill coefficient \(h\). The enzyme identities are indicated on the top of the panels. Values of \(k_{cat}\) were fit to the equation \(k_{cat} = k_{cat,0} + (k_{cat,0} - k_{cat,M})/\left[1 + (K_m(M)]\right)^n\) (with \(n\) equal to 1 and 2 for \(cp\) and \(cp\)PPase, respectively) where \(k_{cat,0}\) and \(k_{cat,M}\) are the limiting values of \(k_{cat}\) at zero and infinite Mg\(^{2+}\) concentrations, respectively, and \(K_m\) is the metal binding constant. The lines for \(K_m\) and \(K_c\) show the best fits for Equation 6. The horizontal dotted lines (\(h = 1\)) mark the boundary between positive and negative cooperativity. \(K_m\) values are measured in terms of the MgPPi complex. Error bars represent S.E.

TABLE 1

| Enzyme | \(k_{cat,0}\) S\(^{-1}\) | \(k_{cat,M}\) S\(^{-1}\) | \(K_m\) M\(^{-1}\) | \(K_m\) dependence | \(K_m\) dependence | \(K_m\) dependence |
|-------|-----------------|-----------------|-------------|-----------------|-----------------|-----------------|
|       |                 |                 |             | (\(K_m\))\(_0\) | (\(K_m\))\(_M\) | (\(K_m\))\(_M\) |
| dhPPase | NA\(^a\) | 320 ± 20 | NA | 47 ± 0.5 | 60 ± 20 | 0.6 ± 0.2 |
| cpPPase | 370 ± 90 | 820 ± 120 | 2 | NA | 22 ± 2 | NA |
| cpPPase | 27 ± 5 | 1310 ± 60 | 1.05 ± 0.05\(^b\) | <10 | 81 ± 3 | ND\(^c\) |
| ePase | 70 ± 14 | 1300 ± 60 | 1.05 ± 0.05\(^b\) | 47 ± 2.5 | 240 ± 10 | 0.14 ± 0.02 |
| cpPPase | 27 ± 5 | 1310 ± 60 | 1.05 ± 0.05\(^b\) | <10 | 81 ± 3 | ND\(^c\) |
| ePase | 70 ± 14 | 1300 ± 60 | 1.05 ± 0.05\(^b\) | 47 ± 2.5 | 240 ± 10 | 0.14 ± 0.02 |

\(^a\) NA, not attendant.
\(^b\) Estimated assuming binding of two metal ions with the same binding constant (see Fig. 3 legend for details).
\(^c\) ND, not determined.

rapid substrate-dependent change in enzyme oligomeric structure upon dilution into the assay medium.

The Hill coefficient \(h\) as well as the ratio \(4K_m/\left[2K_m\right]\) for \(dhPPase\) were similarly greater than 1 across the entire 0.05–20 mM Mg\(^{2+}\) concentration range (Fig. 3) but decreased at low Mg\(^{2+}\) concentrations, indicating a decrease in the degree of cooperativity. This transition resulted from changes in \(K_m\) but not \(K_M\), which did not vary significantly (Fig. 3). The dependence of \(K_m\) on Mg\(^{2+}\) concentration obeyed Equation 6 with the parameters \(K_m0\) and \(K_mM\) replaced by the respective parameters \(K_m0\) and \(K_mM\). The Michaelis constants at zero, and infinite Mg\(^{2+}\) concentration. The fitted \(K_m0\) and \(K_mM\) and \(K_m\) values were 4.7 \(\mu\)M, 60 \(\mu\)M, and 0.6 mM, respectively (Table 1). The measured value of \(k_{cat}\) for \(dhPPase\) showed little dependence on Mg\(^{2+}\) concentration (0.05–20 mM) and was in the range of 300–350 S\(^{-1}\).

A similar kinetic analysis was conducted with three other CBS-PPases. The results are summarized in Fig. 3 and Table 1. With \(cp\)PPase, \(K_m1\) was virtually independent of Mg\(^{2+}\) concentration, whereas \(K_m2\) increased at high [Mg\(^{2+}\)]. As a consequence, Mg\(^{2+}\) exerted an opposite effect on cooperativity compared with \(dhPPase\), and the Hill coefficient dropped to slightly below unity at high [Mg\(^{2+}\)]. With \(cp\)PPase and \(ePase\), both \(K_m1\) and \(K_m2\) changed with [Mg\(^{2+}\)] concentration but in opposite directions. Consequently, the degree of cooperativity increased with increasing [Mg\(^{2+}\)]. The largest effect was observed with \(cp\)PPase, which exhibited \(h\) values in the range of 1.0–1.8 depending on Mg\(^{2+}\) concentration.

The measured value of \(k_{cat}\) was nearly constant over the entire [Mg\(^{2+}\)] range for \(ePase\) but changed 2-fold for \(cp\)PPase and 50-fold for \(cp\)PPase (Fig. 3). For the latter enzyme, \(k_{cat}\) depended on the second power of Mg\(^{2+}\) concentration (see Fig. 3 legend), indicating that two metal ions are required for the transition from \(k_{cat,0}\) to \(k_{cat,M}\).

The above analyses were based on the assumption that the \(k_{cat}\) value for \(E_2S\) is half that for \(E_2S_2\) in Scheme 1.
zero activity for the E₂S species increased the sum of the squared residuals by severalfold, indicating significantly worse fits. In contrast, assuming equal activities of E₂S and E₂S₂ decreased the quality of the fits to a lesser extent, making this case a likely alternative. However, if the ratio of the $k_{cat}$ values for E₂S and E₂S₂ was treated as an adjustable parameter, its best fit value was generally closer to 0.5 than 1.0. Importantly, the values of $K_{m1}$, $K_{m2}$, and their ratio showed essentially the same trends with the $k_{cat}$ ratio equal to 0.5 and 1, indicating positive cooperativity, which decreased at low [Mg$^{2+}$] in both cases. We thus conclude that Scheme 1 is the simplest scheme consistent with the data because any value of the $k_{cat}$ ratio greater than 0.5 implies additional negative catalytic cooperativity because of diminished activity of one of the active sites in the dimeric enzyme.

Kinetic Behavior of dhPPaseΔCDC—Remarkably, dhPPaseΔCDC, which is devoid of the regulatory unit, demonstrated Michaelian kinetics in the presence of 5 mM Mg$^{2+}$ and over the entire 0.1–20 mM Mg$^{2+}$ concentration range as indicated by a 4$K_{m1}/K_{m2}$ value for Scheme 2 and a Hill coefficient close to 1 (Fig. 4 and Table 1). The single microscopic $K_m$ value describing dhPPaseΔCDC kinetics showed little variation in the metal concentration range used, and its values (3.8–5.0 µM) were markedly less than $\sqrt{K_{m1}K_{m2}}$ (the geometric mean of the two $K_m$ values) values for dhPPase (Fig. 4).

The value of $k_{cat}$ for dhPPaseΔCDC was 100–120 s$^{-1}$ over the 0.1–20 mM Mg$^{2+}$ concentration range. The somewhat lower activity of dhPPaseΔCDC compared with dhPPase may indicate that deletion of the regulatory part of the protein caused some distortions in the active site.

Cooperativity of Nucleotide Binding to dhPPase—Fig. 5 shows the concentration dependence of nucleotide effects on activity measured at fixed substrate (MgPPi) and Mg$^{2+}$ concentrations. Micromolar concentrations of AMP and ADP caused inhibition in most cases, whereas ATP acted as an activator. Qualitatively similar effects of these nucleotides were reported previously for the CBS domain-containing mtPPase (18). The major new finding here is that, in most cases where the size of the effect allowed quantitative analysis, the curves were poorly described in terms of a simple 1:1 binding model (Equation 3) but obeyed Equation 2 for cooperative binding to two sites with concomitant activation or inhibition. The quality of the fit was relatively insensitive to the value of $A_{1/2}$ in Scheme 1, but better fits were generally observed with $A_{1/2} = (A_0 + A_\infty)/2$ compared with the limiting cases of $A_{1/2} = 0$ or $A_\infty$. Importantly, most curves generated $K_{N1}$ values greater than $K_{N2}$ regardless of the $A_{1/2}$ value used and Hill coefficients greater than 1 (Table 2). Thus, nucleotide binding also exhibited positive cooperativity. In only two pairs (cpPPase/ADP and ePPase/ATP) did the analysis indicate insignificant cooperativity.
Interestingly, ADP did not affect cnPPase activity, and AMP was equally ineffective with elPPase (Fig. 5). Activity was nucleotide-insensitive in these enzyme-nucleotide pairs over the entire 0.05–20 mM Mg2+ concentration range.5 To test whether the nucleotides bound to enzyme in these cases, we performed competition experiments (Fig. 6A). In both cases, a “functionally silent” nucleotide modulated the activating effect of ATP, indicating that the former is able to bind. Moreover, although ineffective by itself, AMP synergistically enhanced the effect of ADP on elPPase, shifting the inhibition profile to lower ADP concentrations and decreasing the activity value extrapolated to infinite ADP concentration (Fig. 6B). This profile shift is explained by a 3-fold lower \( K_{N1} \) value for ADP in the presence of 100 \( \mu \text{M} \) AMP. The reaction medium contained 50 \( \mu \text{M} \) MgPPi and 5 mM Mg2+ in all cases. Activities without any nucleotide were taken as 100%. The lines show the best fits to Equation 2, rel. relative.

5 V. A. Anashkin, unpublished observations.

### TABLE 2

Parameters describing nucleotide effects on four CBS-PPases as derived from Figs. 5 and 6 and similar data measured at different substrate concentrations

| Nucleotide | [MgPPi] | \( v_0/v_0^* \) | \( K_{N1} \) | \( K_{N2} \) | \( \Delta K_{N1}/K_{N2} \) | \( \sqrt{K_{N1}/K_{N2}} \) | \( h \) |
|------------|---------|----------------|------------|------------|----------------|----------------|--------|
| AMP        | 5       | 0.13 ± 0.02    | 3 ± 2      | 0.3 ± 0.2  | 40 ± 30        | 0.95 ± 0.05    | 1.75 ± 0.14 |
| AMP        | 50      | 0.037 ± 0.001  | 4.8 ± 0.7  | 0.41 ± 0.04| 48 ± 10        | 1.39 ± 0.04    | 1.68 ± 0.04 |
| AMP        | 400     | 0.035 ± 0.006  | 6.7 ± 1.5  | 0.5 ± 0.1  | 53 ± 13        | 1.83 ± 0.05    | 1.74 ± 0.05 |
| ADP        | 5       | 0.23 ± 0.01    | 0.38 ± 0.05| 0.14 ± 0.02| 11 ± 3         | 0.24 ± 0.01    | 1.51 ± 0.05 |
| ADP        | 50      | 0.09 ± 0.01    | 0.99 ± 0.09| 0.90 ± 0.08| 4.4 ± 0.8      | 0.95 ± 0.02    | 1.34 ± 0.03 |
| ADP        | 400     | 0.05 ± 0.01    | 1.4 ± 0.1  | 2.1 ± 0.2  | 2.6 ± 0.4      | 1.72 ± 0.04    | 1.23 ± 0.04 |
| ATP        | 5       | 3.2 ± 0.1      | 2.0 ± 0.3  | 0.63 ± 0.08| 13 ± 3         | 1.14 ± 0.02    | 1.55 ± 0.04 |
| ATP        | 50      | 2.5 ± 0.2      | 2.3 ± 0.8  | 1.5 ± 0.1  | 6 ± 2          | 1.9 ± 0.2      | 1.45 ± 0.13 |
| ATP        | 400     | 2.1 ± 0.1      | 4 ± 1      | 1.7 ± 0.5  | 9 ± 5          | 2.2 ± 0.1      | 1.48 ± 0.10 |

\( v_0^* \) and \( v_0 \) are activities extrapolated to infinite concentration of the variable nucleotide and measured in the absence of any nucleotide, respectively.

ND, not determined.

* Measured in the presence of 20 \( \mu \text{M} \) ATP.

** Measured in the presence of 100 \( \mu \text{M} \) ATP.

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highly cooperative: the first nucleotide molecule binds quite weakly, but the mean binding constant \( \frac{1}{K_{N1}K_{N2}} \) is comparable with that of the other nucleotides. It is unlikely that the extraordinarily high values of \( 4K_{N1}/K_{N2} \) and \( h \) for AMP, which are close to their limiting values for two-site binding (27), are somehow associated with the ADP present in the system because the latter nucleotide exhibited quite low binding cooperativity with \( ePPase \) (Table 2).

The above analysis of nucleotide effects was conducted at a half-saturating substrate concentration. Because the substrate itself also binds cooperatively, the apparent nucleotide binding cooperativity may result from nucleotide effects on substrate binding. Therefore, measurements were extended to low and high substrate concentrations at which the enzyme (\( dhPPase \)) predominantly exists in the substrate-free form or as enzyme-substrate complex, respectively. As Table 2 makes clear, the cooperativity of AMP and ATP binding as characterized by \( 4K_{N1}/K_{N2} \) and \( h \) did not depend significantly on substrate concentration and showed only a moderate change in the case of ADP binding. Therefore, the observed cooperativity of nucleotide binding is not induced by bound substrate or by modulation of substrate binding. In contrast, the relative final activity at infinite nucleotide concentration \( (v_N/v_0) \) increased with increasing substrate concentration in all cases, whereas the mean binding affinity \( (\sqrt{K_{N1}K_{N2}}) \) significantly decreased.

The \( Mg^{2+} \) dependence of AMP, ADP, and ATP binding to \( dhPPase \) and of AMP binding to \( cpPPase \) was determined (Fig. 7). These enzymes and nucleotides were chosen because they demonstrate profound effects on activity, allowing accurate analysis of the binding curves. Although quite large, the effect of ATP on \( ePPase \) was not included in this analysis because ATP chelates \( Mg^{2+} \) appreciably (25) at the high ATP concentrations at which this enzyme operates (Fig. 5), introducing uncertainty in free \( Mg^{2+} \) concentration at the lower values in this range. Varying \( Mg^{2+} \) concentration in the assay changed \( K_{N1} \) and \( K_{N2} \) values for AMP binding to \( dhPPase \) and \( cpPPase \) in opposite directions (Fig. 7). As a result, the ratio \( 4K_{N1}/K_{N2} \), the Hill coefficient \( h \), and hence the degree of cooperativity increased with increasing \( Mg^{2+} \) concentration. With ATP, \( K_{N1} \) but not \( K_{N2} \), was \( Mg^{2+} \)-dependent, resulting in a similar although smaller effect. With ADP, both binding constants changed in parallel without affecting \( 4K_{N1}/K_{N2} \) or \( h \). Notably, \( dhPPase \) activity at saturating ATP concentration \( (470 \pm 10 \text{ s}^{-1}) \) was independent of \([Mg^{2+}]\).

The dependences of \( K_{N1} \) and \( K_{N2} \) on \( Mg^{2+} \) concentration \([M]\) obeyed Equation 6 derived for a simple model that assumes that metal binding to a site characterized by the dissociation constant \( K_M \) shifts the dissociation constant for the enzyme-nucleotide complex from \((K_N)_0 \) to \((K_N)_M \). The fitted values of these parameters are summarized in Table 3. Interestingly, \( K_M \) values were essentially independent of the nucleotide and enzyme used and correlated well with the \( K_M \) value derived from substrate hydrolysis kinetics for \( dhPPase \) (and other PPases) (Table 1). Moreover, in all cases, the \( K_M \) values derived from \( K_{N2} \) dependence were greater than those derived from the \( K_{N1} \) dependence; in both cases, their ratio was not much different from 4, the value expected for non-cooperative metal binding to two identical sites.

Effects of Nucleotides on Kinetic Cooperativity—Table 4 summarizes nucleotide effects on hydrolysis kinetics measured at a \( Mg^{2+} \) concentration of 5 mM. The general trend was that nucleotides decreased kinetic cooperativity as characterized by \( h \) val-
Nucleotide-regulated Soluble Pyrophosphatases

TABLE 3
Kinetic parameters for nucleotide inhibition derived from the dependencies of $K_{N1}$ and $K_{N2}$ on $Mg^{2+}$ concentration (Fig. 7)
Substrate (MgPPi) concentration was fixed at 50 $\mu$M.

| Nucleotide | $K_{N1,0}$ | $K_{N1,1}$ | $K_{N1,2}$ | $K_{N2,0}$ | $K_{N2,1}$ | $K_{N2,2}$ |
|------------|------------|------------|------------|------------|------------|------------|
| dhPPase    | AMP        | 1.7 ± 0.1  | 190 ± 100  | 0.6 ± 0.1  | 7 ± 3      | 0.9 ± 0.3  | 1.4 ± 0.9  |
|            | ADP        | 0.73 ± 0.03| 1.73 ± 0.06| 0.5 ± 0.1  | 1.02 ± 0.04| 3.2 ± 0.2  | 1.3 ± 0.2  |
|            | ATP        | 0.83 ± 0.07| 2.9 ± 0.2  | 0.3 ± 0.1  | NA        | NA        | NA         |
| cpPPase    | AMP        | 0.6 ± 0.1  | 26 ± 11    | 0.3 ± 0.1  | 5.6 ± 0.2  | 2.4 ± 0.2  | 1.6 ± 0.5  |

*NA, not attendant.

FIGURE 8. Activities of CBS-PPases as a function of [ADP]/[ATP] ratio. The reaction medium contained 50 $\mu$M MgPPi, 5 mM Mg$^{2+}$, and different concentrations of ADP and ATP such that their sum was 0.5 mM. Activity in the presence of 0.5 mM ATP was taken as unity in all cases. Error bars represent S.D.

The organisms that harbor the CBS-PPases used in this study are Gram-positive anaerobic bacteria belonging to the phylum Firmicutes (D. hafniense, C. novyi, C. perfringens) or Actinobacteria (E. lenta). Three of them (C. novyi, C. perfringens, and E. lenta) are pathogenic and are found in the human intestinal tract. D. hafniense is found in soil and is capable of reductive dechlorination of chlorophenols (31). In three of these bacteria (excluding C. perfringens), PP metabolism depends on soluble and membrane-bound PPases (32), which convert metaboli-
Cooperativity of nucleotide binding is not caused by bound an 81-fold change in AMP level if it bound non-cooperatively.

**CBS domain-containing proteins.** Positive cooperativity was observed in the general case and is a combination of six individual rate constants in the case of PPase (36). Therefore, a change in PPase activity relates to nucleotide binding and serves to amplify the activity to the regulatory mechanism. The first level of cooperativity relates to enzyme interaction with substrate as characterized by the kinetic cooperativity in CBS-PPases, which bind all three nucleotides. However, the possibility that Mg$^{2+}$ changes the kinetic cooperativity in cpPPase in the opposite direction (Fig. 3) and did not affect ADP binding cooperativity in dhPPase (Fig. 7). Similar values of the Mg$^{2+}$ binding constant, $K_{M}$, derived from measurements of hydrolysis kinetics (Table 1) and nucleotide effects (Table 3) suggest that a single site controls both cooperativity types. Notably, the structure of the AMP-bound regulatory portion of cpPPase crystallized in the presence of Mg$^{2+}$ contains no bound divalent metal ion (19). In contrast, canonical family II PPases, which represent the catalytic part of CBS-PPases, bind three or four Mg$^{2+}$ ions in the absence of substrate (5) and up to five metal ions in its presence (6). All metal ions are located in the active site, but one of them is not absolutely required for activity (6). This last activity-modulating site is thus the most likely candidate for the site that modulates cooperativity in CBS-PPases.

All $K_{N}$ values are reported here in terms of total nucleotide concentrations without regard to complexation with Mg$^{2+}$. Based on the Mg$^{2+}$ binding constants (see “Experimental Procedures”), 99.8% of ATP, 98% of ADP, and 70% of AMP exist as magnesium complexes at the highest Mg$^{2+}$ concentration used (20 mM). Thus, it is not unlikely that nucleotide-bound Mg$^{2+}$ modulates enzyme-nucleotide interactions and therefore contributes to the dependences shown in Fig. 7. However, several lines of evidence rule out the possibility that Mg$^{2+}$ binding to nucleotides is the sole cause of the Mg$^{2+}$ effects on $K_{N}$ values. As already mentioned, two molecules of AMP bind to cpPPase as free nucleotides (19). If recalculated in terms of free AMP, $K_{N1}$ and $K_{N2}$ values for dhPPase and cpPPase (Fig. 7) would decrease in parallel (up to 3-fold at the highest Mg$^{2+}$ concentration), resulting in a minor effect on the AMP binding pattern. The crystal structures of the cpPPase fragment (19) suggest that other nucleotides form major contacts with the enzyme through their AMP moieties. Therefore, free and Mg$^{2+}$-complexed ADP or ATP are likely to have equal or similar affinities for enzyme. This interpretation is consistent with similar mean $K_{N}$ values ($\sqrt{K_{N1}K_{N2}}$) for AMP, ADP, and ATP observed for dhPPase and cpPPase, which bind all three nucleotides. However, the possibility that Mg$^{2+}$ binding to ADP and ATP contributes to their binding patterns in Fig. 7 cannot be excluded.

The structural basis for the effects of the regulatory element on the active site and for the modulation of these effects by bound nucleotides still remains to be determined for PPase as well as for other CBS domain-containing enzymes and transporters. Our results pose further questions in this context. Because the two AMP molecules bound per dimeric regulatory fragment of cpCBS-PPase are in proximity to each other (19), one would expect negative rather than positive cooperativity in their binding. Indeed, AMP analog binding was shown to exert
negative cooperativity in mtfPPase (21). The reversal of nucleotide binding cooperativity in Pase observed in the present study may reflect plasticity of CBS domains and is consistent with the known variability of their amino acid sequences (10). The available crystallographic data show significant opening of the CBS domain interface in the activator (AP₃A) complex of cp_PPase compared with that in the inhibitor (AMP) complex (19). Lucas et al. (38) observed an open-to-closed conformational change in the CBS domain pair of Methanocaldococcus jannaschii protein MJ0100 upon successive binding of adenosine derivatives. Quite recently, leong et al. (39) reported a dramatic AMP-induced structural change in Arabidopsis thaliana protein CBSX2.

The concept of “autoinhibition” was introduced vis-à-vis CBS domains by Janosik et al. (40) to explain cystathionine β-synthase activation by S-adenosyl-l-methionine as well as its limited proteolysis and thermal denaturation. According to this concept, the CBS domain insert acts as an “internal inhibitor,” sensitizing proteins with catalytic or transport functions to structural changes caused by nucleotide binding and permitting either activation or further inhibition. CBS domain-containing PPases provide strong support for this theory. Thus, deletion of the regulatory domains in dlp_PPase favorably changed its $K_{cat}$ value (Table 3). A similar deletion in cystathionine β-synthase increased $k_{cat}$ but not $K_m$ (41, 42). The concept of autoinhibition is also consistent with the diminished activity (1–3 orders of magnitude) of CBS domain-containing PPases compared with their close homologs lacking CBS domains (5) as well as their ability to be activated by ATP. Many mutations in the CBS domains also activate mtfPPase, and some even reverse the effects of nucleotide from inhibition to activation (43), resembling the effects of mutations in cystathionine β-synthase (44) and AMP-dependent protein kinase (45, 46).

Unlike other known CBS domain-containing proteins, dlPPase, cnPPase, and cp_PPase contain a DRTGG domain insert within the CBS domain pair. The results of the present work do not shed light on the function of this extra domain in these (and most other) CBS-PPases as we did not observe any fundamental differences in the behavior of these PPases compared with that of eLPase, which has no DRTGG domain in its structure.

In summary, CBS-PPase presents a unique case, demonstrating in one protein the diverse regulatory capabilities of CBS domains as sensors of cell energy status. The regulation of this enzyme is based on a complex interplay among the effects of nucleotides, substrate, and $\text{Mg}^{2+}$ on binding cooperativity. Further work is clearly needed to fully elucidate this interplay and determine its importance for other CBS domain proteins.

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