In the past decade, many two-component monooxygenases have been identified and studied (1, 2). The enzymes of this class, which are comprised of reductase and monooxygenase components, are involved in a wide variety of reactions, including monooxygenation of p-hydroxyphenylacetate (3–5), phenol (6, 7), chlorophenol (8), 2,4,6-trichlorophenol (9), pyrrole-2-carboxylate (10), styrene (11, 12), p-nitrophenol (13), nitritotriacetate (14), ethylenediaminetetraacetic acid (EDTA) (15), alkane sulfonate (16), and aldehyde (bacterial luciferase) (17, 18). These oxygenases are also involved in the biosynthesis of the antibiotics, actinorhodin (19, 20), pristinamycin IIA (21), and valinomycin (22), as well as the halogenation in the biosynthesis of rebeccamycin (23, 24).

The reductase components of these enzymes generate reduced flavin to be used in the monooxygenation reactions catalyzed by the oxygenases. The mechanism by which the reactive reduced flavin is transferred between protein components is one of the most interesting research questions in this field and is the main focus of this paper, in which p-hydroxyphenylacetate hydroxylase (HPAH)† from Acinetobacter baumannii was used as a model for this study.

HPAH catalyzes the hydroxylation of p-hydroxyphenylacetate (HPA) to form 3,4-dihydroxyphenylacetate (DHPA), and the enzyme has been identified and studied from four bacterial species; Pseudomonas putida (25, 26), Pseudomo-
nas aeruginosa (27), Escherichia coli W (4, 28), and A. baumannii (3, 29–31). Interestingly, although catalyzing the same reaction, the enzymes fall into two distinct types: the reductases from E. coli and P. aeruginosa are dimers of 18–22 kDa monomers, while those from A. baumannii and P. putida are dimers of 34–36 kDa monomers (1, 3–5). The reductase component (C1) of HPAH from A. baumannii is isolated with one flavin mononucleotide (FMN) bound per subunit; its function is to provide the reduced FMN (FMNH–) required for C2 to hydroxylate HPA (3, 29). Our previous studies showed that the binding of HPA allosterically stimulates the reduction of the C1-bound FMN by reduced nicotinamide adenine dinucleotide (NADH) (30).

This property has not been observed in the HPAH reductases from either E. coli or P. aeruginosa (27). The oxygenases from E. coli and P. aeruginosa are dimers of 59 kDa monomers, while the oxygenase from A. baumannii is a tetramer of 47 kDa monomers. When amino acid sequences of the reductases and oxygenases are compared, it is clear that the two types of HPAH do not have a common origin (29). Although the different types of HPAH catalyze the same reaction, they have significant differences in the details of the mechanisms involved.

Thermodynamic data, including redox potentials and Kd values, imply that apoC1 binds to FMNH– less tightly than to oxidized FMN and that the binding to FMNH– is even weaker in the presence of HPA (30). Our earlier investigations in which the rate constants for each step of catalysis were determined for the reactions of C2 with FMNH–, HPA, and O2 [Figure 1 (31)] demonstrated preferential random-order interactions, with C2 binding FMNH– first and then usually O2, followed by HPA. C2 was shown to bind tightly to FMNH– (Kd for C2–FMNH– ~ 1.2 μM), contrasting with its much weaker binding to oxidized FMN (Kd = 250 ± 50 μM) (31). The rate of the reaction to form the C(4a)-hydroperoxy-FMN (the hydroxylation intermediate) when C2 is 20 μM, FMNH– is 10 μM, and O2 is 120 μM is 185 s–1 either when a mixture of C2 and FMNH– is mixed with a solution containing O2 or when a solution containing C2 and O2 is mixed with one containing FMNH–. Therefore, the formation of the C2–FMNH– complex is very rapid, much greater than the 185 s–1 rate to form the C(4a)-hydroperoxy-FMN (31) (Figure 1). This suggests that it is possible that C1-bound FMN, after being released by NADH, can be released from C1 and transferred to the active site of C2 via diffusion without requiring specific protein–protein interactions between the two components.

Flavin transfer between components in this enzyme class has been investigated in several systems, and the answers obtained are diverse, e.g., simple flavin diffusion for E. coli HPAH (4, 28), complex formation for bacterial luciferase (32,33) and alkyl sulfonate monoxygenase (34,35), and a combination of diffusion and complex formation for styrene monoxygenase (12). However, no quantitative information as to how simple diffusion can be effective in these systems

\[ \text{FMNH}_2 \rightarrow \text{FMNH} \rightarrow \text{FAD} \rightarrow \text{FMN} \]

\[ \text{FAD} \rightarrow \text{FMN} \]

\[ \text{FMN} \rightarrow \text{FMNH} \]

\[ \text{FMNH} \rightarrow \text{FMNH}_2 \]

\[ \text{K} = \frac{[\text{FMNH}_2][\text{FAD}]}{[\text{FMN}][\text{FAD}]} \]

\[ \text{K} = \frac{[\text{FMNH}][\text{FAD}]}{[\text{FMN}][\text{FAD}]} \]

\[ \text{K} = \frac{[\text{FAD}]}{[\text{FMN}]} \]

\[ \text{K} = \frac{[\text{FMNH}_2]}{[\text{FMNH}] + [\text{FAD}]} \]

\[ \text{K} = \frac{[\text{FAD}]}{[\text{FMNH}]} \]

\[ \text{K} = \frac{[\text{FMNH}_2]}{[\text{FMNH}] + [\text{FAD}]} \]

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\[ \text{K} = \frac{[\text{FMNH}_2]}{[\text{FMNH}] + [\text{FAD}]} \]

\[ \text{K} = \frac{[\text{FMNH}][\text{FAD}]}{[\text{FMN}][\text{FAD}]} \]

\[ \text{K} = \frac{[\text{FAD}]}{[\text{FMN}]} \]

\[ \text{K} = \frac{[\text{FMNH}_2]}{[\text{FMNH}] + [\text{FAD}]} \]

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\[ \text{K} = \frac{[\text{FMNH}_2]}{[\text{FMNH}] + [\text{FAD}]} \]

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\[ \text{K} = \frac{[\text{FMNH}_2]}{[\text{FMNH}] + [\text{FAD}]} \]

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\[ \text{K} = \frac{[\text{FMNH}_2]}{[\text{FMNH}] + [\text{FAD}]} \]

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\[ \text{K} = \frac{[\text{FMNH}_2]}{[\text{FMNH}] + [\text{FAD}]} \]

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\[ \text{K} = \frac{[\text{FAD}]}{[\text{FMN}]} \]
cloned, expressed, and prepared as previously described (3, 29). The concentration of C1 was estimated from the extinction coefficient, ε 280 = 12.8 × 10³ M⁻¹ cm⁻¹, and the concentration of C2 was estimated from the value (on the basis of the amino acid sequence) ε 280 = 5.67 × 10⁴ M⁻¹ cm⁻¹ (30, 31).

Spectroscopic Studies. UV–vis absorbance spectra were recorded with a Hewlett-Packard diode array spectrophotometer (HP 8453A) or a Shimadzu 2501PC spectrophotometer. Fluorescence measurements were carried out with a Shimadzu RF5301PC spectrofluorometer. All spectral instruments were equipped with thermostated cell compartments.

Rapid Reaction Experiments. All reactions were carried out in buffer A: 50 mM sodium phosphate buffer at pH 7.0 and 4 °C, unless otherwise specified. When appropriate, solutions were made anaerobic by equilibrating with oxygen-free argon as described earlier (31). Rapid kinetics measurements were performed with Hi-Tech Scientific stopped-flow spectrophotometers, model SF-61DX in double-mixing mode or model SF-61SX in single-mixing mode. Optical path lengths of the observation cells were 1 cm. The stopped-flow instrument was made anaerobic by flushing the flow system with an anaerobic solution consisting of 400 μM glucose oxidase, 1 mg/mL glucose oxidase (15.5 unit/mL), and 4.8 μg/mL catalase in 50 mM sodium phosphate buffer at pH 7.0. An anaerobic glucose/glucose oxidase solution was allowed to stand in the flow system overnight, and the flow unit was then thoroughly rinsed with anaerobic buffer prior to experiments.

Apparent rate constants from kinetic traces were calculated from exponential fits using KinetAsyst 3 software (Hi-Tech Scientific, Salisbury, U.K.) or program A (written at the University of Michigan by Rong Chang, Jung-yen Chiu, Joel Dinverno, and David P. Ballou). FMN (alone or in complex with either C1 or C2) was reduced anaerobically in a tonometer with dithionite (~5 mg/mL stock solution in 100 mM potassium phosphate buffer at pH 7.0) delivered anaerobically from a syringe prior to reactions with O2 in the stopped-flow apparatus. Reduction reactions were monitored spectrophotometrically. Buffer solutions containing various concentrations of oxygen were prepared by bubbling solutions with mixtures of certified nitrogen and oxygen.

Gel-Filtration Chromatography To Assess Affinities of C1 to FMN and FMNH₂. The affinity of the C1 active site for FMN was investigated by passing solutions of 32 μM oxidized C1 in the presence of 200 μM HPA through a Sephadex G-25 gel-filtration column (1.4 × 11 cm) previously equilibrated with 50 mM sodium phosphate buffer at pH 7.0, 1 mM dithiothreitol (DTT), and 0.5 mM EDTA at 25 °C. This procedure was used to separate free FMN from enzyme-bound forms. In other experiments, 32 μM reduced C1, either with or without 200 μM HPA was similarly passed through the same Sephadex G-25 gel-filtration column. NADH (1 mM) was included in the equilibration and elution buffers to maintain the FMN in the reduced state. The flow rate for elution was 1.5 mL/min. Fractions of 1.5 mL were collected from the column and left at room temperature for about 30 min until the reduced flavin (colorless upon elution) became fully oxidized (yellow) before analysis. Each of the fractions was monitored for protein and FMN concentrations by measuring the absorbance at 280 and 450 nm, respectively.

Table 1: Initial Rates of Product (DHPA) Formation Measured in Steady-State Kinetics

<table>
<thead>
<tr>
<th>[FMN] (μM)</th>
<th>initial rate (μM s⁻¹)</th>
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<th>initial rate (μM s⁻¹)</th>
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<tbody>
<tr>
<td>0.0</td>
<td>3.9</td>
<td>3.7</td>
<td>3.5</td>
</tr>
<tr>
<td>1.0</td>
<td>3.8</td>
<td>3.7</td>
<td>3.6</td>
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<td>8.0</td>
<td>3.4</td>
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<td>3.4</td>
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<tr>
<td>16.0</td>
<td>3.2</td>
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Steady-State Kinetics in the Presence of C1 and C2. Most steady-state kinetics studies of the two-component flavin-dependent hydroxylases have been carried out with flavin in great excess over the two enzymes. If the reductase was used to have a high catalytic turnover rate compared to the oxygenase (which is usually the case), the excess free reduced flavin produced could react with O2 and lead to considerable H2O2 (and even superoxide). This would result in the overall hydroxylation being poorly coupled to the use of NADH. We have carried out experiments here to test whether excess FMN is necessary or whether the FMN that is bound to the reductase (C1) as isolated is sufficient for efficient catalysis. Steady-state survey kinetics using 2 μM C1, 4 μM C2, 200 μM NADH, 120 μM HPA, and various concentrations of free FMN were monitored by measuring the NADH consumption at 340 nm in the stopped-flow instrument. It was observed that when the added FMN concentration was decreased from 1 to 0.01 μM, the initial rate only changed from 21.7 to 8.8 μM/s, less than 3-fold (data not shown). We also carried out reactions containing 100 μM HPA, 80 μM NADH, and various concentrations of free FMN (1, 2, 4, 8, and 16 μM) to test whether product formation was stimulated by added FMN. The initial rate of formation of DHPA was measured using 3,4-dihydroxycinnamylactate dioxygenase to convert the product to the highly colored 5-carboxymethyl-2-hydroxymuconate semialdehyde, which is easily detected (see Table 1) (3). The addition of free FMN did not increase the initial rate of product formation. In fact, the apparent rate seemed to be slightly smaller at high concentrations of free FMN (Table 1). It might be noted that at these higher FMN concentrations this apparent loss of activity could be due to the reduction of added FMN by the reductase, which would result in decreases of absorbance at 420 nm that could partially counter the decrease as a result of the formation of the product. Thus, at this range of concentrations of C1 and C2, external FMN is not needed for catalysis. The flavin initially bound by C1 can function as both a cofactor and substrate for C2; FMN is reduced on C1, transferred to C2, where it reacts with O2.

RESULTS

Steady-State Kinetics in the Presence of C1 and C2. Most steady-state kinetics studies of the two-component flavin-dependent hydroxylases have been carried out with flavin in great excess over the two enzymes. If the reductase was used to have a high catalytic turnover rate compared to the oxygenase (which is usually the case), the excess free reduced flavin produced could react with O2 and lead to considerable H2O2 (and even superoxide). This would result in the overall hydroxylation being poorly coupled to the use of NADH. We have carried out experiments here to test whether excess FMN is necessary or whether the FMN that is bound to the reductase (C1) as isolated is sufficient for efficient catalysis. Steady-state survey kinetics using 2 μM C1, 4 μM C2, 200 μM NADH, 120 μM HPA, and various concentrations of free FMN were monitored by measuring the NADH consumption at 340 nm in the stopped-flow instrument. It was observed that when the added FMN concentration was decreased from 1 to 0.01 μM, the initial rate only changed from 21.7 to 8.8 μM/s, less than 3-fold (data not shown). We also carried out reactions containing 100 μM HPA, 80 μM NADH, and various concentrations of free FMN (1, 2, 4, 8, and 16 μM) to test whether product formation was stimulated by added FMN. The initial rate of formation of DHPA was measured using 3,4-dihydroxycinnamylactate dioxygenase to convert the product to the highly colored 5-carboxymethyl-2-hydroxymuconate semialdehyde, which is easily detected (see Table 1) (3). The addition of free FMN did not increase the initial rate of product formation. In fact, the apparent rate seemed to be slightly smaller at high concentrations of free FMN (Table 1). It might be noted that at these higher FMN concentrations this apparent loss of activity could be due to the reduction of added FMN by the reductase, which would result in decreases of absorbance at 420 nm that could partially counter the decrease as a result of the formation of the product. Thus, at this range of concentrations of C1 and C2, external FMN is not needed for catalysis. The flavin initially bound by C1 can function as both a cofactor and substrate for C2; FMN is reduced on C1, transferred to C2, where it reacts with O2.
and HPA to effect the hydroxylation, and then returns to C₁ to begin another catalytic cycle.

A steady-state kinetics study with no added FMN was therefore carried out in buffer A using the stopped-flow spectrophotometer. Assays contained 2 μM C₁, 4 μM C₂, and various concentrations of HPA (10, 20, 40, 80, 100, 120, 140, and 160 μM). Because the reaction of C₂−FMNH² with O₂ is very fast, the concentration of O₂ was kept at 240 μM (aerobic), which is likely to be well in excess of Kₘ for O₂.

The solutions containing the enzymes and various concentrations of HPA were mixed with 100 μM NADH, and the reactions were monitored at 340 nm. All concentrations are given after mixing. It was found that HPA concentrations greater than 120 μM resulted in substrate inhibition, as described previously (31) and discussed below. Therefore, subsequent assays used HPA at 100 μM and various concentrations of NADH (10, 20, 40, 80, 100, 120, and 140 μM). Inhibition occurred at NADH concentrations ≥ 100 μM. The apparent Vₘₐₓ was determined to be 4 μM/s under these conditions, yielding an apparent Kₘₚₐₓ ~ 2 s⁻¹ (data not shown). This value is similar to the apparent rate constant of 1.8 s⁻¹ calculated from single-turnover experiments for the last phase of the reaction of C₂−FMNH² with these concentrations of HPA and O₂. In this last step, the C(4a)-hydroxy-FMN product formed from the hydroxylation reaction (via kₘ in Figure 1) loses H₂O (kₖ in Figure 1) to form oxidized FMN (31). As described below, high concentrations of HPA trap the C(4a)-hydroxy-FMN to retard its dehydrogenation to form FMN. Thus, the dehydrogenation of C₂ is the rate-limiting step of the overall hydroxylation reaction.

The coupling of product formation to NADH utilization was tested under the same conditions as used above for the steady-state kinetics study. The DHPA product was quantified by high-performance liquid chromatography (HPLC) analysis using NADH as the limiting substrate, and HPA was 120 μM. These experiments showed that an average of 86% of the NADH was used in the hydroxylation of HPA (data not shown). This value is similar to that determined from single-turnover reactions (~80%) when based on the amount of reduced flavin used in the reactions (31).

Overall, the results from the steady-state kinetics studies allow us to conclude that (a) no extra FMN (above that in the reductase) is required for effective catalysis, (b) the rates of product formation are nearly optimal under these conditions, and (c) FMNH⁻ is transferred from the reductase to the oxygenase, with very little being oxidized while free in solution (i.e., there is good coupling).

Separation of FMN(H⁺) from C₁ under Equilibrium Conditions. Experiments were carried out to examine qualitatively whether FMNH⁻ binds less tightly to C₁ than does FMN and whether HPA has further effects on this putative differential binding. G-25 Sephadex gel-filtration chromatography (Figure 2) was used to separate free from bound FMN in both oxidized and reduced forms, as described in the Materials and Methods. The peak centered at fraction 6 contains all of the protein and therefore indicates the amount of flavin that is bound to C₁. Fractions 9–20 contain free flavin. When oxidized C₁ (no HPA present) was chromatographed, essentially all of the FMN eluted with the protein (data not shown), which is consistent with the known tight binding of FMN to C₁ (Kₖ = 6 nM at 4 °C (30)). The solid line of Figure 2 represents a chromatogram of oxidized C₁ in the presence of 200 μM HPA. Most of the FMN eluted with C₁ (holoenzyme), but a small amount of free FMN was detected, consistent with its slightly weaker binding in the presence of HPA [Kₛ₈ = 38 nM at 4 °C (30)]. The dashed line is a chromatogram of reduced C₁ in the absence of HPA, which is also shown in Figure 2. The solution containing C₁ plus 200 μM HPA, a dotted line for HPA-free reduced C₁, and a line with filled circles for reduced C₁ plus 200 μM HPA.

Evidence that C₂ Binds FMNH⁻ More Tightly than C₁. Efficient transfer of flavin between the two components during catalysis requires that FMNH⁻ binds more tightly to C₂ (the hydroxylase) than to C₁ (the reductase) and that FMN binds more tightly to C₁. A solution containing 16 μM C₁ in buffer A was placed in a cuvette and made anaerobic. C₁ was reduced with a slight excess of sodium dithionite to ensure that the flavin remained reduced during the experiment; reduction of C₁ was monitored spectrophotometrically. When C₂ was tipped in from a side arm to be at a final concentration of 25 μM, the reduced flavin spectrum became perturbed as shown in Figure 3A. This perturbed spectrum is very similar to that when C₂ was added to a solution of free FMNH⁻ (Figure 3B), strongly suggesting that it is due...
to the binding of FMNH\(^-\) to C\(_2\). This conclusion implies that C\(_1\) binds FMNH\(^-\) with a \(K_d\) \leq\) that for C\(_1\). Note that the solid lines in parts A and B of Figure 3 are not exactly the same because only \(\sim 80\%\) of FMNH\(^-\) was transferred from C\(_1\) to C\(_2\) under the conditions used in Figure 3A (see explanation in the next experiment).

**Reaction of Reduced C\(_1\) Plus C\(_2\) with Oxygen.** If, in an anaerobic mixture of C\(_1\) and C\(_2\), most of the FMNH\(^-\) is bound to C\(_2\), such a mixture would be expected to react with O\(_2\) essentially the same as when C\(_1\) is not present. Buffer A containing 15 \(\mu\)M C\(_1\) and 25 \(\mu\)M C\(_2\) was stoichiometrically reduced with sodium dithionite (see the Materials and Methods) and then mixed at 4 °C in a stopped-flow spectrophotometer with buffer A containing various concentrations of oxygen (Figure 4). Concentrations are given in the caption as after mixing in the stopped-flow instrument. Reactions were monitored at 385 and 458 nm (Figure 4A). Kinetic analysis indicated that the reaction consisted of three phases, with the first phase being complete by \(\sim 0.02\) s, the second phase being complete by \(\sim 5\) s, and the third phase being complete by \(\sim 200\) s. The first phase is shown by an increase of absorbance at 385 nm with no change at 458 nm, indicating the formation of C(4a)-hydroperoxy-FMN with no formation of oxidized FMN. A plot of \(k_{obs}\) for this phase was linearly dependent upon the oxygen concentration, with a second-order rate constant of \(1.2 \pm 0.1 \times 10^6\) M\(^{-1}\) s\(^{-1}\) (inset of Figure 4A). These results are nearly identical to those for the formation of C(4a)-hydroperoxy-FMN in the reaction of a complex of C\(_2\) and FMNH\(^-\) with oxygen in the absence of C\(_1\) [Table 2 (31)]. This finding is consistent with the binding experiment above, suggesting that

![Figure 3: Evidence for binding FMNH\(^-\) from C\(_1\) or free in solution to the C\(_2\) active site. (A) C\(_1\) (16 \(\mu\)M) was reduced by a slight excess of sodium dithionite in an anaerobic cuvette, and its spectrum is shown as the dotted line. Then, C\(_2\) (25 \(\mu\)M final) was added from the side arm into the cuvette, and the spectrum (...) was recorded. (B) This experiment was carried out in the same manner as in A, but 16 \(\mu\)M of free FMNH\(^-\) was used instead of reduced C\(_1\).](image)

![Figure 4: Reaction of reduced C\(_1\) plus C\(_2\) with oxygen. (A) Anaerobic solution of 15 \(\mu\)M reduced C\(_1\) and 25 \(\mu\)M C\(_2\) was mixed in a stopped-flow spectrophotometer with buffer A containing various concentrations of oxygen. The solid and dotted lines are traces measured at 385 and 458 nm, respectively, using oxygen concentrations of 0.13, 0.31, 0.61, and 1.03 mM (right to left). The inset shows the observed rate constants for the formation of C(4a)-hydroperoxy-FMN as a function of the oxygen concentration (the phase from 0 to 10 ms), from which a second-order rate constant of \(1.2 \pm 0.1 \times 10^6\) M\(^{-1}\) s\(^{-1}\) was calculated. (B) Data from similar experiments as in A using 1.03 mM O\(_2\) were recorded at selected wavelengths between 310 and 550 nm. A spectrum of the C(4a)-hydroperoxy-FMN intermediate (line with filled circles) was obtained by plotting the values from the traces at 10 ms. The spectrum has an absorption peak at 380 nm. The spectrum of reduced C\(_1\) plus C\(_2\) is shown as the dashed line, while that of the final oxidized species with FMN bound to C\(_1\) is shown as the solid line.](image)
alone, because most of the FMNH\(^-\) initially bound to C\(_1\) had been transferred to C\(_2\) before the reaction with O\(_2\). Kinetic traces of the reaction described in Figure 4A using 1.03 mM oxygen were monitored at 5–10 nm intervals in the region of 315–550 nm. Values at the reaction time of 0.01 s were used to construct a spectrum of the C\(_4\)-hydroperoxy-FMN intermediate (Figure 4B). The spectrum (filled circles in Figure 4B) is typical for spectra of C(4a)-oxygenated flavin adducts (38–41) and the one observed during the reaction of C\(_2\)–FMNH\(^-\) with oxygen (31). Note that because of the incomplete transfer of FMNH\(^-\) from C\(_1\) to C\(_2\), by 10 ms under these conditions, the absorbance of the intermediate in Figure 4B is less than that from the reaction of C\(_2\)–FMNH\(^-\) with oxygen (31). The results above suggested that the presence of HPA makes the transfer of FMNH\(^-\) from C\(_1\) to C\(_2\) more favorable. To test this hypothesis, anaerobic buffer A containing 16 \(\mu\)M reduced C\(_1\), 25 \(\mu\)M C\(_2\), and 400 \(\mu\)M HPA was mixed in a stopped-flow spectrophotometer with buffer A containing 400 \(\mu\)M HPA and various concentrations of oxygen. The reaction monitored at 370 nm shows four phases (Figure 5A). At the highest oxygen concentration (the left most trace), the first phase was an increase of absorbance during the dead time until about 4 ms and the rate was dependent upon the oxygen concentration (1.2 ± 0.3 \(\times\) 10\(^6\) M\(^{-1}\) s\(^{-1}\)). This second-order rate constant is the same as that for the formation of C(4a)-hydroperoxy-FMN in the absence of HPA during the oxidation of C(4a)-FMNH\(^-\) (31) and for the first phase of the reaction of the reduced C\(_1\) plus C\(_2\) with oxygen (Figure 4A). The second phase in Figure 5A consists of a large increase in absorbance from \(~1.7–22\) s\(^{-1}\) and this rate was also dependent upon the oxygen concentration (5.0 ± 0.3 \(\times\) 10\(^4\) M\(^{-1}\) s\(^{-1}\)). This value is essentially the same as that previously measured for the reaction of oxygen with C\(_2\)–FMNH\(^-\) in complex with HPA [Figure 1 and Table 2 (31)]. Therefore, the first and second phases are likely to be due to the formation of C\(_2\)–C(4a)-hydroperoxy-FMN intermediates in the HPA-free and HPA-bound forms. The third phase consists of a small decrease in absorbance between 0.1 and 1 s and was independent of the oxygen concentration, with an apparent rate constant of \(~17–22\) s\(^{-1}\) (Figure 5A), similar to the hydroxylation step occurring in the reaction of oxygen with C\(_2\)–FMNH\(^-\) in complex with HPA [Figure 1 and Table 2 (31)]. The fourth phase is seen as an absorbance decrease at 370 nm and a large increase at 458 nm (dotted-line traces), with an apparent rate constant of 1.1 s\(^{-1}\). This phase was due to the dehydration of C(4a)-hydroxy-FMN to yield oxidized FMN and is dependent upon the concentration of HPA: when HPA is at 2 mM, an apparent rate constant of

<table>
<thead>
<tr>
<th>Table 2: Kinetic and Thermodynamic Constants for the Reactions of Reduced C(_1) Plus C(_2) with O(_2) and Reactions of C(_2)–FMNH(^-) Alone (31)</th>
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<td>kinetic or thermodynamic constants</td>
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<td>(k_5), formation of C(4a)-hydroperoxy-FMN(^a)</td>
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<td>(k_5), formation of C(4a)-hydroperoxy-FMN in the presence of HPA(^b)</td>
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<td>(K_0), for the reduced enzyme/HPA complex(^c)</td>
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<td>(K_0), for the enzyme-C(4a)-hydroperoxy-FMN/HPA complex(^c)</td>
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<td>(k_5), for the isomerization of the C(_2)–C(4a)-hydroperoxy-FMN/HPA complex</td>
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<td>(k_5), for hydroylation(^c)</td>
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<td>(k_5), for product release(^c)</td>
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<td>(k_5), dehydroporation (and release of FMN)(^d)</td>
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\(^a\) Kinetic constants correspond to the labels in Figure 1. \(^b\) Values from single-mixing stopped-flow experiments. \(^c\) Values from double-mixing stopped-flow experiments. \(^d\) Strongly dependent upon the HPA concentration.
0.35 s\(^{-1}\) is observed. The presence of 2 mM HPA traps hydroxy-FMN, so that the loss of H\(_2\)O to form oxidized FMN occurs slowly (31).

Spectra of two intermediates were calculated from a set of traces similar to those shown in Figure 5A, recorded at 5–10 nm intervals from 315 to 550 nm. The first spectrum was plotted from the values observed at 4 ms. The second intermediate was calculated using absorbance values at the reaction time of 0.08 s, corrected for full formation using rate constants for the oxygen reaction of 54 s\(^{-1}\) (the O\(_2\) reaction under these conditions in the presence of HPA), the hydroxylation step of 22 s\(^{-1}\), and the dehydration step of 1.1 s\(^{-1}\). The line with open circles represents a spectrum of C(4a)-hydroperoxy-FMN formed from that fraction (\(\sim 25\%\)) of the enzyme that does not have HPA bound; it is similar in character to that shown in the previous section obtained without HPA (Figure 4B). The line with closed circles represents a spectrum of C(4a)-hydroperoxy-FMN formed by the end of the second phase, and it represents the spectrum of the HPA-bound form of the enzyme (which reacts more slowly with oxygen than the free enzyme (31)) plus that of any enzyme without HPA bound. It appears that the presence of HPA slightly shifts the spectrum to shorter wavelengths compared to that in Figure 4 (375 nm versus 380 nm). Overall, these results are very similar to those obtained without C1 present (31) and are also consistent with most of the C1-bound FMNH\(^{-}\) having been transferred to C2 in the presence of HPA before being mixed with O\(_2\).

**Determination of the Rate Constant for the Transfer of FMNH\(^{-}\) from C1 to C2 in the Absence and Presence of HPA.** From our earlier study (31), the rate constant for free FMNH\(^{-}\) binding to C2 (Figure 1) was estimated to be \(>10^7\) M\(^{-1}\) s\(^{-1}\). This large rate constant explains why mixing free FMNH\(^{-}\) with an aerobic solution of C2 resulted in the complete formation of C2−C(4a)-hydroperoxy-FMN at the same rate as when preformed C2−FMNH\(^{-}\) was mixed with oxygen [the C(4a)-hydroperoxy-FMN intermediate forms with a second-order rate constant of 1.2 × 10\(^6\) M\(^{-1}\) s\(^{-1}\) (31)]. Because the C(4a)-hydroperoxy-FMN forms very rapidly (see Figure 4A), its appearance can be used to indicate when FMNH\(^{-}\) is transferred from C1 to C2. For example, if the transfer of FMNH\(^{-}\) from C1 to C2 is much faster than the rate of the oxygen-dependent formation of C(4a)-hydroperoxy-FMN, the intermediate should form with the same second-order rate as when C1 is absent (as in the reaction of the C2−FMNH\(^{-}\) complex). On the contrary, if the rate of FMNH\(^{-}\) transfer is slower than the rate of the formation of C(4a)-hydroperoxy-FMN, the observed rate of its formation will indicate the rate of transfer and will not be second-order with respect to O2. A solution of reduced C1 was mixed in a stopped-flow spectrophotometer with solutions of C2 in buffer A containing various concentrations of O2. The reactions were monitored at 380 and 458 nm (Figure 6).

In the absence of HPA, C(4a)-hydroperoxy-FMN formed at only \(\sim 0.35\) s\(^{-1}\), with some dependence on the O2 concentration (380 nm, dotted lines), and this intermediate converted to oxidized FMN at 0.037 s\(^{-1}\) (458 nm, dotted line in Figure 6). Thus, in the absence of HPA, flavin transfer is slow and occurs at only \(\sim 0.35\) s\(^{-1}\). Moreover, because it is so slow, some C1−FMNH\(^{-}\) becomes oxidized by its direct reaction with O2 (note the 458 nm trace, dotted line, out to \(\sim 10\) s). In contrast to the reaction with C1−FMNH\(^{-}\), the reaction of free FMNH\(^{-}\) with C2 under these conditions has a rate of formation of the C(4a)-hydroperoxy-FMN of 185 s\(^{-1}\); this intermediate converts to oxidized FMN very slowly at 0.037 s\(^{-1}\), the same as observed in the above experiment (31).

The presence of 2 mM HPA dramatically changes these reactions. The solid lines in Figure 6 show the results of mixing reduced C1 containing 2 mM HPA with solutions of C2 also containing 2 mM HPA and various concentrations of O2. At 380 nm, the observed rate constants for the formation of the C(4a)-hydroperoxy-FMN (first phase, 0.002–0.2 s) were all less than those for the reaction of C2−FMNH\(^{-}\) with O2 (see Figure 4A and Table 2) and were not linearly dependent upon the oxygen concentration. The k\(_{\text{obs}}\) values for this phase ranged from 55 to 74 s\(^{-1}\) as O2 varied from 0.13 to 1.03 mM. This indicates that the rate of transfer of FMNH\(^{-}\) from C1 to C2 occurs at a rate that is \(\geq 74\) s\(^{-1}\). The conversion of the C(4a)-FMN adduct to oxidized FMN under these conditions occurs at \(\sim 0.35\) s\(^{-1}\), as shown by the trace recorded at 458 nm (solid line trace of 458 nm in Figure 6). In this reaction, hydroxylation proceeded at \(\sim 20\) s\(^{-1}\) to form C(4a)-hydroxy-FMN (k\(\text{j}\) in Figure 1), which has almost the same spectrum as C(4a)-peroxy-FMN. However, as discussed above, the presence of 2 mM HPA traps the hydroxy-FMN, so that the loss of H\(_2\)O to form oxidized FMN occurs at only 0.35 s\(^{-1}\). Note that in the previous section this rate was 1.1 s\(^{-1}\) when less HPA (400 mM) was present (31). At very low concentrations of HPA, the rate of this process is extrapolated to be \(\sim 8\) s\(^{-1}\), as indicated in Figure 1. It is a coincidence that, with this concentration of HPA (2 mM), the formation of FMN after hydroxylation (0.35 s\(^{-1}\)) is the same as the rate of transfer of FMNH\(^{-}\) from C1 to C2 in the absence of HPA.

We also used a double-mixing stopped-flow approach to measure the rate constant for transfer of FMNH\(^{-}\) from C1 to C2 in the absence of HPA. First, reduced C1 (16 \(\mu\)M) was mixed with C2 (25 \(\mu\)M) anaerobically and aged for various periods. Then, buffer that contained oxygen (0.5 mM after mixing) was added in the second mix, and the reactions were monitored at 380 nm to detect the rapid formation of C(4a)-hydroperoxy-FMN (Figure 7). At this concentration of O2, the reaction with C2−FMNH\(^{-}\) completely forms C2−C(4a)-hydroperoxy-FMN by 10 ms (31), so that its formation provides a good measure of the amount of FMNH\(^{-}\) that has
resulting in a greater formation of C₂ from C₁ at the same rate as the transfer of FMN in the rapid phase. A control experiment without C₂ to C₁ (dotted line of Figure 7) showed that no C₂ has been transferred at any aging time after mixing. Figure 7 shows that with longer aging times there were larger absorbance increases by 10 ms, demonstrating that, when C₁–FMNH₂ was incubated with C₂ for increasing periods of time, more FMNH₂ was transferred from C₁ to C₂, resulting in a greater formation of C₂–C(4a)-hydroperoxy-FMN in the rapid phase. A control experiment without C₂ present showed that no C₂–C(4a)-hydroperoxy-FMN was formed and the flavin simply reoxidized and then rebound to C₁ [dotted line of Figure 7 (30)]. A plot of the absorbance increases at 380 nm that had occurred at the reaction time of 10 ms versus the aging time (inset of Figure 7) represents the kinetics of transfer of FMNH₂ from C₁ to C₂ in the absence of HPA. A half time of 1.97 s was observed, corresponding to a rate constant of 0.35 s⁻¹, the same as obtained in the single mix experiment above. In the presence of HPA, the transfer is too fast (∼80 s⁻¹) to resolve by our double-mixing technique.

**Use of Cyt c To Measure the Release of FMNH₂ from C₁.** Results presented in previous sections have shown that FMNH₂ bound to C₁ can be efficiently transferred to C₂ and react at the C₂ active site; there was no indication that C₁ and C₂ need to interact to facilitate this transfer (Figures 2–7). Moreover, the dissociation of FMNH₂ from C₁, which allows for its binding to C₂, was clearly shown to be facilitated by HPA binding to C₁ (Figure 6). In the following experiments, we used the reduction of cyt c by C₁–FMNH₂ as a second approach to examine whether FMNH₂ is released from C₁ at the same rate as the transfer of FMNH₂ to C₂ described in the previous section. It is known that the reduction of cyt c by free FMNH₂ is very fast and occurs in one-electron steps (37, 42–44). Reduction of cyt c by free FMNH₂ was carried out as a control experiment as described in Figure 8. Reduction of cyt c by FMNH₂ can be monitored by the increase of absorbance at 550 nm because of the appearance of the α band of reduced cyt c (inset of Figure 8). Although the reaction (solid line) exhibits complex kinetics, the reduction of cyt c by FMNH₂ was complete by 20 ms. About 50% of the change occurred in the dead time of the stopped-flow instrument, and the remainder occurred with an observed rate constant of approximately 500 s⁻¹ (the second electron transfer). As expected, the change of absorbance up to 20 ms is equivalent to the reduction of 2 molecules of cyt c/molecule of FMNH₂ present. The slower phases are probably due to the presence of a small excess of dithionite initially in the FMNH₂ solution and possibly to some photochemistry. In the following experiments, it was assumed that cyt c would not react rapidly with FMNH₂ bound to C₁ (this was found to be true, see below), so that the kinetics of the reduction of cyt c by reduced C₁ in the presence and absence of HPA would indicate the rates of dissociation of FMNH₂ from the C₁ active site. Successively, the same reduction of cyt c was investigated in the presence of C₂. If a specific interaction between reduced C₁ and C₂ occurs during transfer, the kinetics of cyt c reduction by C₁ would be significantly affected by the presence of C₂.

When cyt c was mixed with reduced C₁ under the same conditions as in the control experiment (above), cyt c was reduced with an observed rate constant of only ∼0.35 s⁻¹ (dashed line in Figure 8), the same rate constant as observed for the FMNH₂ transfer in the absence of HPA (Figures 6 and 7). This suggests that, in the absence of HPA, the release of FMNH₂ from C₁ occurs at 0.35 s⁻¹ and is rate-limiting in the transfer of FMNH₂ from C₁ to C₂. This result also implies that the C₁-bound FMNH₂ does not react rapidly with cyt c; the rate of the reaction is likely to be much less than 0.35 s⁻¹.

When the same experiment was carried out again but in the presence of 2 mM HPA, a very rapid phase of reduction of cyt c occurred in the dead time, typical of that of free flavin (about 40% of the total change), implying that ∼40% of the FMNH₂ was not bound to C₁ when HPA was present. This step was followed by further reduction of cyt c with an observed rate constant of ∼80 s⁻¹ (the filled-circle line in Figure 8 up to 40 ms). Thus, the bound flavin reacted with
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**FIGURE 9:** Reduction of cyt c by reduced C₁ in the presence of C₂. All concentrations given are after mixing, and solutions contained 2 mM HPA. Experiments were carried out using a stopped-flow spectrophotometer. The reduction of cyt c was monitored at 550 nm. Reduced C₁ (8 µM) was mixed with air-saturated buffers containing oxidized cyt c (25 µM) and various concentrations of C₂ (16, 40, 100, and 245 µM, upper to lower solid line traces). A control experiment in which the same concentration of reduced C₁ and HPA was mixed with cyt c without C₂ under the same conditions is shown as the dotted line.

cyt c at ~80 s⁻¹, which is similar to the rate of transfer of FMNH⁻ from C₁ to C₂ as discussed above. Note that, in the presence of HPA, a larger portion of FMNH⁻ was free than when HPA was absent, which is consistent with HPA increasing the affinity between C₁ and FMNH⁻ as indicated in the gel-filtration experiments (Figure 2). Thus, we conclude that the release of FMNH⁻ from C₁ is the process that governs the rate of flavin transfer from C₁ to C₂ and HPA stimulates this rate by ~200-fold; diffusion and binding to C₂ are very fast.

In the next experiment, we examined whether specific interactions between C₁ and C₂ are required for the dissociation of FMNH⁻ from C₁. If the efficient transfer of FMNH⁻ from C₁ to C₂ in the presence of HPA requires specific interactions between the two proteins, C₂ would be expected to exclude or alter the kinetics of cyt c reduction by reduced C₁. A solution containing reduced C₁ and 2 mM HPA was mixed with an air-saturated solution of cyt c and various concentrations of C₂, and the reduction of cyt c by the FMNH⁻ from C₁ was monitored at 550 nm (Figure 9). Because free FMNH⁻ binds very rapidly to C₂, there was no need in this experiment to carry out the reaction anaerobically. The reaction traces in Figure 9 show that the rate constants observed (~80 s⁻¹) for the reduction of cyt c by FMNH⁻ from C₁ (reactions up to ~40 ms) were independent of the concentration of C₂ and the same as that previously determined for the dissociation of FMNH⁻ from C₁ in the absence of C₂ (Figure 8). This is because the reduction of cyt c is limited by the dissociation process, and C₂ has no influence on the dissociation of FMNH⁻ from C₁. However, the amplitude of the absorbance change decreased with increasing concentrations of C₂, indicating that C₂ competed with cyt c for FMNH⁻ that had been released from C₁ at ~80 s⁻¹. It should be emphasized that, even with the concentration of C₂ much greater than that of cyt c (up to ~10-fold or 245 µM), the same rate of reduction of cyt c was still observed (the lowest line, Figure 9), indicating that there is no specific protein interaction between C₁ and C₂ to facilitate this FMNH⁻ transfer process.

**Oxidation of Free FMNH⁻ with and without Superoxide Dismutase.** The traces shown in Figure 10 demonstrate that,

when FMNH⁻ reacts with O₂ at 10.5% saturation (half of the concentration of aerobic solutions or ~130 µM after mixing), very little FMNH⁻ is oxidized in the first 0.1 s. Only 3 and 6% of the FMNH⁻ was oxidized by 0.1 s with and without superoxide dismutase present, respectively. This experiment demonstrates that, in the presence of HPA, FMNH⁻ can dissociate from C₁ with a rate of 80 s⁻¹ and rapidly bind to C₂ with a rate of ≥10⁷ M⁻¹ s⁻¹ before its reaction with O₂ can occur.

**DISCUSSION**

Two main findings come from the work described here. First, it is demonstrated that the C₁/C₂ system can function efficiently with no added flavin; the flavin on C₁ as isolated is sufficient for nearly full activity of the system. Second, it has been demonstrated that the reduced flavin (FMNH⁻) can be readily transferred from C₁ to C₂ via simple diffusion without requiring any protein–protein interactions. Moreover, the kinetic measurements reported here show quantitatively why simple diffusion can function successfully in catalysis.

Results from steady-state kinetics experiments (Table 1) have shown that, when C₁ and C₂ are in the micromolar range, exogenous flavin added has very little influence on the rate of product formation. The thermodynamics of binding for reduced and oxidized FMN to each of the components are appropriate for fostering the passing of FMN in a ping-pong fashion between the components of HPAH during catalysis. C₁ binds tightly to oxidized FMN with a Kₐ of 0.006 µM in the absence of HPA and a Kₐ of 0.038 µM in its presence (30). The gel-filtration experiments (Figure 2) showed that virtually all of the FMN eluted with C₁, which is consistent with the above Kₐ. In contrast, FMNH⁻ binds to C₂ significantly less tightly than does FMN; a considerable fraction eluted separately from C₁ when HPA was not present, and about 50% eluted separately from C₁ in its presence. The Kₐ for the C₁−FMNH⁻ complex is 1.2 µM, while that for the C₂−FMN complex is ~250 µM (31). These results demonstrate that in the presence of both proteins FMN binds more tightly to C₁ and FMNH⁻ binds more tightly to C₂, as required for efficient catalysis.

After completion of the hydroxylation reaction on C₂, the resulting oxidized FMN dissociates from C₂ and rebinds to
C1. Our stopped-flow results (discussed below) and the final spectrum showing characteristics of the C1-bound flavin after completion of the single-turnover reaction of C1 and C2 (Figures 4B and 5B) demonstrate that FMN in its appropriate oxidation states can be readily transferred between C1 and C2; added free flavin is not needed in this system for efficient hydroxylation to occur (Figures 3–6). Given the Kd values of the functional complexes, C2−FMNH− and C1−FMN (1 and 0.038 μM, respectively), efficient passage of the flavin between the components is feasible if the concentration of C2 is higher than 1 μM and the concentration of C1 is higher than 0.05 μM. Previously, Louis et al. have estimated the concentrations of the oxygenase and reductase components of HPAH in E. coli cells to be ∼122 and ∼15 μM, respectively (28). Thus, if C1 and C2 are present in quantities as low as one-tenth of those of the E. coli system, its Kd values for FMN(H+) would permit each component to efficiently sequester the oxidized or reduced FMN released from the other and extra FMN added would not significantly improve the efficiency of the system.

It should be noted that, in our earlier report on steady-state kinetic studies of C1 and C2, the rate of product formation was influenced by the concentration of free flavin (3). In retrospect, it is likely that this was due to the low concentrations of C1 and C2 (1–10 nM) used in the assays, causing the enzymes to exist mostly in the free form; adding extra flavin enhanced the formation of the C2−FMNH− complex and therefore increased the rate of hydroxylation (3). It can be noted that most steady-state studies previously reported for two-component flavin-dependent hydroxylase systems used very small concentrations of enzymes, which, as noted above, are not likely to mimic those found in the bacteria.

In mixtures of reduced C1 plus C2, FMNH− is primarily bound to C2, especially when HPA is present. This was shown by several experiments. First, when reduced C1 and C2 are added together at equilibrium, the spectrum of the FMNH− changes to that of the flavin bound to C2 (Figure 3). Second, reactions in which O2 is mixed with solutions containing reduced C1 plus C2 (in the absence or presence of HPA; Figures 4 and 5) are very nearly the same as corresponding reactions in which O2 is mixed with solutions containing C2 prebound to FMNH− or when solutions of C2 and O2 react with FMNH− (31). Table 2 compares kinetic and thermodynamic constants for the reactions of O2 with C2 in the presence and absence of reduced C1. All parameters for the reaction of oxygen with reduced C1 plus C2 are very similar to those for C2−FMNH− alone, consistent with the FMNH− being primarily on C2. The overall conclusion is that, in mixtures of reduced C1 and C2, FMNH− initially bound to C1 will be transferred to the C2 active site.

The binding of free FMNH− to C2 must be faster than the autoxidation of FMNH− if simple diffusion is to be effective in transferring FMNH− from C1 to C2; this would avoid wasteful auto-oxidation of the flavin during the transfer. Additionally, the regulation of the reduction and release of FMNH− from C1 by HPA would also prevent the reductase from becoming a good oxidase. Our previous results have shown that FMNH− binds to C2 very rapidly (we estimate these reactions to be ≥500 s−1 when C2 is 25 μM) (31). When reduced C1 was reacted with a mixture of C1 and O2 (no pre-equilibration), the reactions were highly dependent upon the presence of HPA (Figure 6). In the absence of HPA, the reaction of a mixture of C2 and 130 μM O2 with reduced C1 to form the C(4a)-hydroperoxy-FMN intermediate on C2 occurred with an observed rate of ∼0.35 s−1. The double-mixing experiment shown in Figure 7 confirmed that, in the absence of HPA, FMNH− was transferred from C1 to C2 in a first-order reaction, with the same rate constant of only 0.35 s−1.

In the presence of HPA, the reaction of reduced C1 with a mixture of C2 and oxygen formed C(4a)-hydroperoxy-FMN and reached a maximum rate of ∼80 s−1. This rate was only partially dependent upon the O2 concentration and was followed by hydroxylation reactions very similar to those observed with C2 alone. At 130 μM O2, the observed rate of formation of the C(4a)-hydroperoxy-FMN was 55 s−1, which is just what would be expected for a consecutive reaction in which dissociation of FMNH− from C1 at ∼80 s−1 was followed by a reaction of oxygen with C2 in complex with FMNH− at ∼185 s−1. The rates of reactions of C1 with O2 in the presence of HPA but in the absence of C2 were ∼80 s−1 because, although FMNH− is released from C1 at ∼80 s−1, in contrast to the reactions involving C2, reactions of O2 with FMNH− are not very fast (see below). Under conditions that assays are usually carried out using very small concentrations of reductase, this slow oxidation of reduced flavin is sufficient to keep up with catalysis.

In this work, we have obtained several lines of evidence that rule out the necessity of C1/C2 complexes being involved in transferring reduced flavin from C1 to C2. No such complex can be isolated by gel filtration under oxidized or reduced conditions in either the presence or absence of HPA. The results from studies using cyt c show that the rates of transfer of flavin are independent of any transient complexes between C1 to C2. Cyt c was reduced by the complex of C1 with FMNH− in the absence of HPA at an observed rate of 0.35 s−1, the same rate as its transfer to C2. In the presence of HPA, the rate was ∼80 s−1 (Figure 8), also in close agreement with the rates of transfer to C2. Finally, we found that C2 can compete with cyt c for FMNH− as it is released from C1; the observed rate for the reduction of cyt c, even in the presence of C2, was still ∼80 s−1. This is not the sum of the two reactions with FMNH− because the rate constant for the release of FMNH− from C1 (80 s−1) is the rate-determining step of this process (Figure 9).

The recently determined X-ray structure of C2 and its complexes with FMNH− and HPA offer an explanation of how C2 can bind FMNH− so rapidly (45). The isosallooxazine of FMNH− binds to C2 at the dimeric interface, and the overall structures of apoC2 and C2−FMNH− are very similar, implying that the C2 structure can efficiently bind FMNH− without requiring any major structural changes or physical interactions with C1.

It is not surprising that diffusion is a suitable mechanism for the transfer of FMNH− to the oxygenases of these two-component flavin-dependent hydroxylases. The reaction of free FMNH− with O2 is autocatalytic, and although the overall reaction is relatively fast (1/kl2 ∼ 0.5 s, depending upon the O2 concentration, temperature, etc.), the initial rate of oxidation is quite slow. The equations below illustrate the important reactions involved in the autocatalytic oxidation of FMNH− (46–48).
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\[
\begin{align*}
FMNH^- + O_2 & \rightarrow \text{slow} \quad FMNH^+ + O_2^- \quad (a) \\
H^+ FMNH^+ + O_2^- & \rightarrow \text{fast} \quad FMNHOOH \quad (b) \\
FMNHOOH & \rightarrow \text{fast} \quad FMN + H_2O_2 \quad (c) \\
FMNH^+ + O_2^- & \rightarrow \text{very fast} \quad FMN + O_2^- \quad (d) \\
H^+ + FMNH^- + FMN & \rightarrow \text{very fast} \quad 2FMNH^+ \quad (e) \\
2H^+ + FMNH^- + O_2^- & \rightarrow \text{fast} \quad FMNH^+ + H_2O_2 \quad (f) \\
FMNH^+ + O_2^- & \rightarrow \text{very fast} \quad FMN + H_2O_2 \quad (g) \\
2O_2^- + 2H^+ & \rightarrow \text{very fast} \quad O_2 + H_2O_2 \quad (h)
\end{align*}
\]

Bruce and colleagues have estimated reaction a to be \(\sim 250 \text{ M}^{-1} \text{s}^{-1}\) at 25 °C, which would give a rate of 0.025 s\(^{-1}\) at 100 \(\mu\text{M}\) \(O_2\). On the other hand, reaction d is very fast. Furthermore, as oxidized FMN is produced, it reacts with FMNH\(^-\) to form more FMNH\(^+\) that can react very fast (reaction e), leading to the observed autocatalysis. Additionally, superoxide, which is produced from reactions a and d, also reacts with various reduced forms of flavin (e.g., reactions f and g) to enhance the autocatalysis (47, 48). Superoxide dismutase, which is present in most organisms, slows the reaction \(\sim 2\)-fold by eliminating reactions d and e while enhancing reaction h, as seen in Figure 10. Figure 10 demonstrates that any reasonably rapid binding process will permit the FMNH\(^-\) to diffuse and bind to the partner oxygenase before it reacts with \(O_2\). For example, a binding rate constant of \(10^6 \text{ M}^{-1} \text{s}^{-1}\) and a concentration of oxygenase of \(10^{-5} \text{ M}\) would give a rate of 10 s\(^{-1}\), which would trap most of the reduced flavin before significant autoxidation occurred. With \(C_2\), the binding rate constant is \(\geq 10^7 \text{ M}^{-1} \text{s}^{-1}\), giving an observed rate \(\geq 100 \text{ s}^{-1}\). As mentioned above, the cellular concentrations of these oxygenase systems are likely to be at least 10 \(\mu\text{M}\) because they are induced to provide a major source of carbon to the bacteria (28).

Most evidence obtained on the mechanism of transfer of reduced flavin between reductases and oxygenases in other two-component systems is inconclusive. Reported kinetic parameters do not firmly establish if rapid diffusion or complex formation is the sole means for transfer of reduced flavin to the oxygenase in each system. A flavin-diffusion transfer model has been favored for \(E. coli\) HPAH because the oxygenase component binds to reduced FAD with a \(K_d\) of 70 nM, whereas it binds to oxidized FAD with a \(K_d\) of 6 \(\mu\text{M}\) (28). The \(E. coli\) HPAH was even able to catalyze the hydroxylation reaction, while both components were separated by a dialysis membrane (4), and it was also shown that the flavin reductases of other systems are able to replace the \(E. coli\) reductase and efficiently generate reduced FAD for its oxygenase (28). These rule out the necessity of specific interactions. However, the results presented here quantitatively show how this is possible and that diffusion is not only possible but also effective.

In the system involving biosynthesis of actinorhodin in \(Streptomyces coelicolor\), an oxygenase component binds to FMNH\(^-\) with a \(K_d\) of 0.39 \(\mu\text{M}\) and to FMN with a \(K_d\) in the range of 19–26 \(\mu\text{M}\), while the reductase binds FMN more tightly than FMNH\(^-\) (20). In styrene monoxygenase from \(Pseudomonas\) sp. VLB120, FAD binds to the oxygenase with a \(K_d\) of 21 \(\mu\text{M}\) and to the reductase with a \(K_d\) of 2.3 \(\mu\text{M}\) (11). These binding parameters are appropriate for the flavin transferring between the reductase and oxygenase proteins during catalysis, suggesting that the flavin in these other enzymes could also be transferred via rapid diffusion. In styrene monoxygenase from \(Pseudomonas\) sp., the oxygenase (SMOA) binds to reduced FAD 137-fold more tightly than to oxidized FAD [\(K_d\) for binding reduced FAD is 0.5 \(\mu\text{M}\) (12)]; however, the simulation of steady-state kinetics results suggested a model where both flavin diffusion and complex formation are involved (12). However, in contrast to our study here, in these other systems, kinetics showing how the oxygenase can bind reduced flavin very rapidly and thus avoid autoxidation have not yet been reported.

Direct channeling or complex formation has been proposed to be the mode of flavin transfer in several systems. Evidence for channeling has been presented by Tu et al. for bacterial luciferase and its reductase systems (18, 49). Data from fluorescence anisotropy measurements of eosin-labeled reductase in the presence of luciferase have provided evidence for complexes (32), and fluorescence energy transfer between luciferase and yellow fluorescent protein (YFP) was observed in the coupled reaction of luciferase and the reductase fused with YFP (33). In alkane sulfonate monoxygenase, a model involving complex formation has been favored on the basis of altered steady-state kinetics of the reductase component when the oxygenase was included (34) and, recently, by detection of a complex by affinity chromatography and cross-linking experiments (35).

Our findings indicate the important role of HPA in regulating the rapid transfer of FMNH\(^-\) between components in the HPAH system. Without HPA, the reduction of C\(_1\)-bound FMN by NADH is 14.7 s\(^{-1}\) (30) and the “off” rate of FMNH\(^-\) from C\(_1\) is 0.35 s\(^{-1}\) (Figures 6–8), rendering the overall production of free FMNH\(^-\) to be rather slow (Figure 11A). In the presence of HPA (Figure 11B), the observed constant rate for the reduction of FMN is 300 s\(^{-1}\) (30), while the observed “off” rate constant is \(\sim 80 \text{ s}^{-1}\) (Figures 6 and 8). Thus, it would appear that HPA, a substrate for the oxygenase component, allosterically influences the reactivity of the reductase and controls the overall rate of production of FMNH\(^-\). In the absence of HPA, the hydroxylation system thereby avoids wasteful production of FMNH\(^-\) to minimize the generation of \(H_2O_2\) and consumption of the cellular reductant, NADH.

When the overall kinetic constants are considered, the rate-limiting step in the reaction of C\(_1\) and C\(_2\) is identified to be the step in which the C(4a)-hydroxy-FMN intermediate dehydrates to return back to the oxidized species (Figure 1). The rate of this step is 1.8 s\(^{-1}\) in the presence of 100 \(\mu\text{M}\) HPA, similar to the value of \(k_{cat}\) (\(\sim 2 \text{ s}^{-1}\)) obtained from the steady-state assays (see the Results). The rate of this step is even less when the concentration of HPA is increased, because the enzyme becomes trapped as the dead-end C(4a)-hydroxy-FMN/HPA complex (31). It is interesting that the transfer of FMNH\(^-\) from C\(_1\) to C\(_2\) (74 s\(^{-1}\); Figure 6) is not remotely rate-limiting in this reaction, although this step requires intermolecular flavin transfer. During steady-state
FIGURE 11: Flavin-diffusion transfer model. (A) In the absence of HPA, the reduction of the C1-bound FMN by NADH occurs at the rate of 14.7 s⁻¹. The reduced FMN is then slowly released from C1 (0.35 s⁻¹), and it diffuses to the C2 active site. (B) In the presence of HPA, C1 undergoes a conformational change that results in a large increase of the reduction rate (300 s⁻¹) and a more rapid dissociation rate of FMNH₂ from C2 (80 s⁻¹).

In conclusion, our study has clearly illustrated that the unstable cofactor, FMNH₂, in the catalytic reactions of a two-component enzyme system, can be transferred between proteins efficiently, with no protein complexes being necessary. With HPAH from A. baumanii, HPA regulates the reduction and release of FMNH₂ from the reductase, enabling the transfer of FMNH₂ by rapid diffusion to the oxygenase.

REFERENCES

Reduced FMN Transfer between the Proteins of HPAH

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